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This is my last volume as editor and the first volume that is published in electronic format only. I would have liked to see JESO continuing to publish hard copy up to and including volume 150, in 2019, but that is not to be. Now, as soon as submitted manuscripts are accepted for publication and edited technically they will be put up on the ESO website for anyone to access freely. No need to wait until the end of the year to see your paper published. Back issues of JESO are also available online, either through the website or the Biodiversity Heritage Library. In time, JESO will have a greater and more useful online presence because we expect to add all past journal articles to the ESO website as individual searchable entities.

Whereas most, if not all, electronic journals have page charges for open access publication JESO does not, so being able to publish without page charges and have anyone access your paper immediately upon posting it on the ESO website should be a pretty good incentive to publish there. Admittedly, people want their paper to appear in "high impact" journals. But not every article is so brilliant or is of such general interest that it will be accepted by the best journals. Rather, most articles are well-substantiated (hopefully) pieces of research that add useful information, whether of interest to few or many, to the overall store of sound scientific knowledge and are therefore worth publishing somewhere. JESO is a good journal to consider. Papers on any aspect of entomology are welcome and the scope need not be restricted to research done in Ontario or on Ontario insects. Taxonomists wishing to publish in JESO should be aware of recent amendments to the International Code of Zoological Nomenclature that detail the mandatory requirements needed to validate new taxa published in e-journals. The essential point is that the work must be registered in ZooBank before it is published. I will work with the technical editor and new editor to determine how this is to be done.

This year's volume is about half as long (75 pp.) as I had hoped but it is better than the low of 55 pages in volume 140. It contains one scientific note, three scientific papers and a review. Please check the ESO website and read/download the articles that interest you. It would be very nice to see the number of published pages increase to a couple of hundred pages each year.

It has been a pleasure to serve as JESO Editor for the past four years. I would like to thank Tom Onuferko at York University, our new technical editor, for his help in preparing the papers for this year's volume, and the associate editors for looking after the review process for manuscripts I passed on to them. Finally, I am pleased to introduce you to Dr. Chris MacQuarrie in Sault Ste. Marie, who has graciously accepted the privilege

and challenge of being the next JESO editor. Please send any manuscripts to him at Chris.
MacQuarrie@nrcan.rncan.gc.ca

I wish all of you the best for the coming year.

John T. Huber
Editor

NEW RANGE RECORDS, AND A COMPARISON OF SWEEP NETTING AND MALAISE TRAP CATCHES OF HORSE FLIES AND DEER FLIES (DIPTERA: TABANIDAE) IN NORTHERN ONTARIO

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Abstract *J. ent. Soc. Ont.* 145: 3–14

Horse flies and deer flies (Diptera: Tabanidae) were surveyed in northern Ontario, Canada in 2011, at 11 sites, and 2012, at 12 sites using Malaise traps and daily sweep netting. A total of 2168 tabanids representing 30 species: 10 *Chrysops*, 18 *Hybomitra*, and two *Tabanus* were collected. Malaise traps caught fewer individuals than sweep netting but more species: 850 tabanids of 28 species, eight of which were not caught by sweep netting. Sweep netting caught 1318 tabanids of 22 species, with two not found in Malaise trap samples. The first record of *Hybomitra osburni* (Hine) in Ontario, and range extensions for several other species are given.

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Introduction

When habitats change, insect populations respond rapidly, up or down, depending on the species characteristics (Niemela *et al.* 1993). These changes occur across a range of temporal and spatial scales, and are unique for each species. This quality makes insect diversity an efficient indicator for monitoring both short and long term environmental changes (Danks 1992). The great variety of habitats occupied by insects in Ontario means that studies that require tracking environmental change can benefit from using some insect group for monitoring that change. For such work, up-to-date distributional data are needed for the insect species of interest.

The eastern "Ring of Fire" region in the eastern area of Northern Ontario contains large deposits of chromium and other minerals (Far North Science Advisory Panel 2010), and anticipated large-scale extraction processes will alter insect diversity and distribution.

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To see the effect of such development, as well as possible effects of changing climates, baseline distributional data for these areas are needed. With this purpose, the Ontario Ministry of Natural Resources (OMNR) started a project in 2009 to survey insect diversity and establish species distributions for Northern Ontario.

A widespread and easy to find and collect group are horse flies and deer flies (Tabanidae). Pechuman *et al.* (1961) compiled the first comprehensive report on the Tabanidae of Ontario. Teskey (1990) provided a more complete treatment of Tabanidae in Canada. Since then, two pictorial keys, one on deer flies (Thomas and Marshall 2009) and one on horse flies (Thomas 2011), added new range information to this group. Further reports of sampling, especially from northern Ontario, continue to add distributional data to our knowledge of Tabanidae (e.g., Beresford 2011).

Here we list the different species of Tabanidae caught in northern Ontario using two different collecting methods and report on range extensions of several of them.

Materials and Methods

We sampled horse flies and deer flies in north-west Ontario in 2011 and north-east Ontario in 2012 (Fig. 1, inset map) at 12 locations each year using two sampling methods, sweep netting and Malaise trapping. Ringrose *et al.* (2013) provided detailed site descriptions and locations. Generally, sampling took place within 1 km of remote field camps that were accessed by helicopter. The 2011 sampling was completed in the western half of Ontario boreal forest within a 150 km radius of the First Nations communities of Big Trout Lake and Sandy Lake. The 2012 sampling occurred in the northeastern part of the province within a 150 km radius of the First Nations community of Fort Albany, Ontario. Sampling dates were from 5 June to 17 July in 2011, and from 5 June to 15 July in 2012.

Tabanids were sampled each day by two methods, Malaise traps (6m trap model no. 2877, BioQuip Products 2321 Gladwick Street, Rancho Dominguez, CA 90220, USA), and sweep netting. The collecting heads of the Malaise traps were filled with 80% denatured ethanol to kill and preserve captured tabanids. These were emptied and replaced each day at 9 pm.

Sweep netting was completed at midday as the surveyor (JLR) walked slowly, sweeping for 5 minutes any Tabanidae that assembled around the researcher. The netted samples were killed by placing the end of the bag in large killing bottles charged with acetone. Specimens were then removed from the net and stored in bottles filled with 80% denatured ethanol. The ethanol in each storage bottle was replaced after 24 hours.

All tabanids were pinned and identified by JLR and DVB using the keys found in Teskey (1990), Thomas and Marshall (2009) and Thomas (2011). The main pinned collection is stored in insect cabinets at Trent University, Biology Department, Peterborough, Ontario. A reference collection of voucher specimens is housed at the Canadian National Collection of Insects, Ottawa.

Analysis

A list of the expected species was produced from the distribution records reported in the keys listed above. For those species that did not have records in northern Ontario,

we reasoned that any species with records that straddled northern Ontario (either east and west, or north and south of the sampling regions) was likely present in northern Ontario. We compared this expected number of species to the predicted number which we calculated using the lognormal distribution method (Preston's method) as described in Ludwig and Reynolds (1988). This approach allows one to predict the number of species present in an area from sampling data. It is based on a general observation that most species are more or less moderately abundant (the middle region of the lognormal distribution), a few species are very abundant (forming the right tail of the lognormal distribution) and a few are very rare (the left tail of the lognormal distribution). In practice, it enables one to predict the number of rare species that are expected but which might have been missed. Parameters for the lognormal model were fitted using the SOLVER function in MICROSOFT EXCEL 2007.

Catch data were analyzed using the online rarefaction calculator from the University of Alberta (http://www.biology.ualberta.ca/jbrzusto/rarefact.php), to determine the effects of collection size on the number of species collected, as well as to compare trapping methods.

Results

Range records and extensions

From our assessment of published range maps, we expected to find 31 species of Tabanidae: 23 with records from across northern Ontario in the regions where we conducted our study (11 *Hybomitra*, 8 *Chrysops*, 2 *Atylotus*, 2 *Tabanus*), and 8 with ranges that straddle our study regions (5 *Hybomitra*, 2 *Atylotus*, and 1 *Haematopota*).

We collected 2168 tabanids from 30 species over the two years: 839 from 24 species in northwest Ontario (2011 sampling), and 1329 from 25 species in northeast Ontario (2012) (Tables I and II, Fig. 1). We found 18 *Hybomitra*, 10 *Chrysops*, and 2 *Tabanus*, but no *Atylotus*, or *Haematopota*.

The expected number of species was calculated to be 26 (lognormal fitted parameters, $\alpha = 0.28$, $S_o = 4.09$, $\chi 2 = 7.93$, $p = 0.34$, d.f.=7) in the western collections (2011) and 28 (fitted parameters, $\alpha = 0.24$, $S_o = 3.73$, $\chi 2 = 9.89$, $p = 0.27$, d.f. = 8) in the eastern collections (2012), and 33 species for the combined data set (fitted parameters, $\alpha = 0.23$, $S_o = 4.27$, $\chi 2 = 7.72$, $p = 0.56$, d.f. = 9).

The three most abundant species caught in the northwest (2011) were *Chrysops excitans* Walker (35%), *Hybomitra epistates* Osten Sacken (21%) and *H. lurida* (Fallén) (19%). In the northeast (2012) the most abundant were *Hybomitra affinis* (Kirby) (33%), *C. excitans* (22%) and *H. lurida* (19%).

New Ontario record

Our collection of three individuals of *Hybomitra osburni* (Hine) (two in 2011 and one in 2012) are the first records of this species in Ontario. This species has been collected in all western provinces and the Yukon Territory (Teskey 1990) but was previously not known to occur east of Manitoba.

TABLE 1. Tabanidae species and number of specimens collected in 2011 and 2012 using Malaise traps and sweep netting, with abundance records.

Species	2011 Malaise	2011 netted	2012 Malaise	2012 netted	Total
Chrysops ater Macquart			1		1
Chrysops cuclux Whitney		1			1
Chrysops dawsoni Philip		2	4	10	16
Chrysops excitans Walker	37	151	245	225	658
Chrysops frigidus Osten Sacken	5		1		6
Chrysops mitis Osten Sacken	11	50	6	5	72
Chrysops niger Macquart			1		1
Chrysops nigripes Zetterstedt	1		1	1	3
Chrysops venus Philip	1				1
Chrysops zinzalus Philip	4		8		12
Hybomitra affinis (Kirby)	7	268	17	62	354
Hybomitra arpadi (Szilady)	8	25	27	25	85
Hybomitra criddlei (Brooks)	1	1			2
Hybomitra epistates Osten Sacken		14	124	153	291
Hybomitra frontalis (Walker)			5	9	14
Hybomitra frosti Pechuman	2				2
Hybomitra hearlei (Philip)			2		2
Hybomitra illota (Osten Sacken)	3	2		1	6
Hybomitra lasiophthalma (Macquart)		21	4	2	27
Hybomitra lurida (Fallén)	59	104	131	121	415
Hybomitra minuscula (Hine)	6	9	3	2	20
Hybomitra nuda (McDunnough)		14			14
Hybomitra osburni (Hine)		2	1		3
Hybomitra pechumani Teskey & Thomas	4	1	8		13
Hybomitra tetrica (Marten)		2	1		3
Hybomitra trepida (McDunnough)		13	19	7	39
Hybomitra typhus (Whitney)		3	8		11
Hybomitra zonalis (Kirby)	2	5	67	6	80
Tabanus marginalis Fabricius			8	1	9
Tabanus vivax Osten Sacken			7		7
Total specimens	151	688	699	630	2168
Total species	15	19	24	15	30

TABLE 2. Tabanidae species listed for each sampling location and date. Only 11 sampling sites are included for 2011 as planned samples were damaged by black bears.

Year	Sampling dates	Longitude (West)	Latitude (North)	Species			
				C. ater	*C. cuclux*	*C. dawsoni*	*C. excitans*
2011	May 31 – June 7	89° 6' 27"	53° 12' 8"				
	May 31 – June 7	89° 40' 42"	54° 25' 49"				
	June 8 – 15	88° 54' 51"	53° 45' 34"				X
	June 8 – 15	92° 1' 43"	54° 9' 29"				
	June 16 – June 23	90° 21' 37"	54° 27' 1"				X
	June 16 – June 23	88° 33' 33"	54° 28' 18"				X
	June 28 – July 5	93° 2' 32"	53° 27' 39"		X	X	X
	June 28 – July 5	94° 13' 38"	52° 49' 27"				X
	July 6 – July 13	91° 49' 8"	52° 27' 37"				
	July 6 – July 13	93° 32' 9"	53° 36' 8"				X
	July 14 – July 21	92° 46' 3"	53° 44' 12"				X
2012	June 5 – June 11	81° 57' 47"	52° 46' 34"				
	June 5 – June 11	82° 39' 13"	51° 55' 53"			X	X
	June 12 – June 18	80° 23' 10"	51° 26' 40"			X	X
	June 12 – June 18	81° 50' 56"	51° 39' 7"				X
	June 19 – June 25	81° 39' 22"	51° 58' 8"				X
	June 19 – June 25	82° 41' 2"	52° 53' 25"				X
	June 26 – July 2	83° 17' 23"	51° 29' 53"			X	X
	June 26 – July 2	82° 49' 1"	52° 28' 26"				X
	July 3 – July 9	89° 6' 27"	53° 12' 8"				X
	July 3 – July 9	89° 40' 42"	54° 25' 49"				X
	July 10 – June 16	88° 54' 51"	53° 45' 34"				X
	July 10 – July 16	92° 1' 43"	54° 9' 29"	X			

TABLE 2 continued...

Year	2011											2012											
Sampling dates	May 31 – June 7	May 31 – June 7	June 8 – 15	June 8 – 15	June 16 – June 23	June 16 – June 23	June 28 – July 5	June 28 – July 5	July 6 – July 13	July 6 – July 13	July 14 – July 21	June 5 – June 11	June 5 – June 11	June 12 – June 18	June 12 – June 18	June 19 – June 25	June 19 – June 25	June 26 – July 2	June 26 – July 2	July 3 – July 9	July 3 – July 9	July 10 – June 16	July 10 – July 16
Species																							
C. frigidus										X													X
C. mitis					X	X	X			X					X		X	X		X	X		X
C. niger																						X	
C. nigripes											X												X
C. venus										X													
C. zinzalus										X	X											X	X
H. affinis					X	X	X			X	X			X	X		X	X	X	X	X	X	X
H. arpadi			X		X	X	X			X					X		X	X	X	X	X	X	
H. criddlei									X	X													
H. epistates							X			X				X	X	X	X	X	X	X	X	X	X
H. frontalis																	X	X		X			
H. frosti											X												
H. hearlei																	X	X					
H. illota			X			X		X									X						

TABLE 2 continued...

Year	2011											2012											
Sampling dates	May 31 – June 7	May 31 – June 7	June 8 – 15	June 8 – 15	June 16 – June 23	June 16 – June 23	June 28 – July 5	June 28 – July 5	July 6 – July 13	July 6 – July 13	July 14 – July 21	June 5 – June 11	June 5 – June 11	June 12 – June 18	June 12 – June 18	June 19 – June 25	June 19 – June 25	June 26 – July 2	June 26 – July 2	July 3 – July 9	July 3 – July 9	July 10 – June 16	July 10 – July 16
Species																							
H. lasiophthalma			X		X		X	X		X				X			X	X					
H. lurida	X		X		X	X	X	X		X		X	X	X	X	X	X		X				
H. minuscula							X				X						X					X	X
H. nuda			X		X		X																
H. osburni			X														X						
H. pechumani							X				X											X	X
H. tetrica			X							X							X						
H. trepida							X	X		X							X	X			X	X	
H. typhus							X												X			X	
H. zonalis					X	X	X			X				X	X		X	X	X		X		
T. marginalis																				X		X	X
T. marginalis																				X	X	X	X

Range extensions

We report nine new northern range records in Ontario. They are: *Chrysops cuclux* Whitney, *C. niger* Macquart, *C. venus* Philip, *Hybomitra criddeli* (Brooks), *H. epistates*, *H. lasiophthalma* (Macquart), *H. pechumani* Teskey & Thomas, *H. tetrica* (Marten), *H. trepida* (McDunnough), and *Tabanus vivax* Osten Sacken. In addition, we provide three new western records of *Hybomitra* for Ontario: *H. minuscula* (Hine), *H. typhus* (Whitney), and *H. frosti* Pechuman.

Gap infill

Chrysops ater Macquart is described as an abundant species having a general northern distribution in Canada south of the tree line (Teskey 1990). Our collection of a single specimen in 2012 is therefore not a surprise; however, there has been little collection in northern Ontario so our collection has filled a gap between previous collecting locations. It is perhaps surprising that it was so rare in our collections. Our records of *Chrysops dawsoni* Philip and *C. frigidus* Osten Sacken are consistent with known ranges.

Chrysops excitans, and *C. mitis* Osten Sacken, and to a lesser extent *C. nigripes* Zetterstedt and *C. zinzalus* Philip, are found in Canada south of the tree line (Teskey 1990), and have been reported from Polar Bear Provincial Park (Beresford 2011). Our records are consistent with these reports.

We caught eight species of *Hybomitra*, consistent with known ranges: *H. affinis*, the most abundant and widely distributed Canadian species of Tabanidae (Teskey 1990; Thomas 2011), *H. arpadi* (Szilady), *H. frontalis* (Walker), *H. hearlei* (Philip), *H. illota*

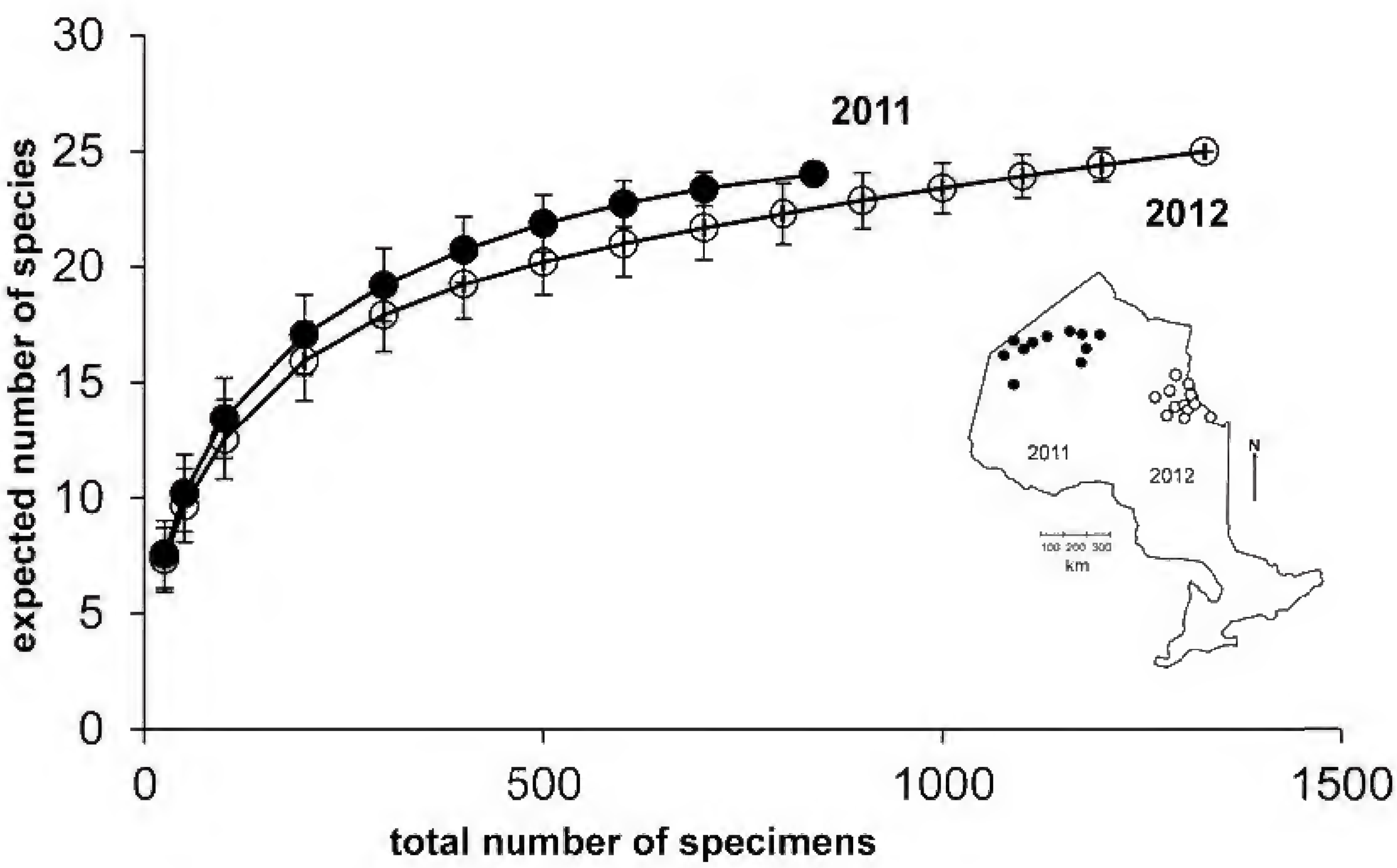

FIGURE 1. Rarefaction analysis showing the expected number of species (y axis) for smaller total catch sizes (x axis), for 2011 (closed circles) and 2012 (open circles). The inset map shows sample locations in both years. Error bars represent standard deviations.

(Osten Sacken), *H. lurida*, *H. nuda* (McDunnough), and *H. zonalis* (Kirby).

Tabanus marginalis Fabricius has been collected from across Canada except on the east coast (Teskey 1990). While the known range encompasses our sampling locations (northern Manitoba and northern Quebec) our records are the northernmost from Ontario.

Trap comparison

The Malaise sampling caught fewer individuals than sweeping yet produced more species. Malaise traps collected 850 specimens of 28 species (151 in 2011 and 699 in 2012); sweep netting collected specimens 1318 of 22 species (688 in 2011 and 630 in 2012) (Fig. 2).

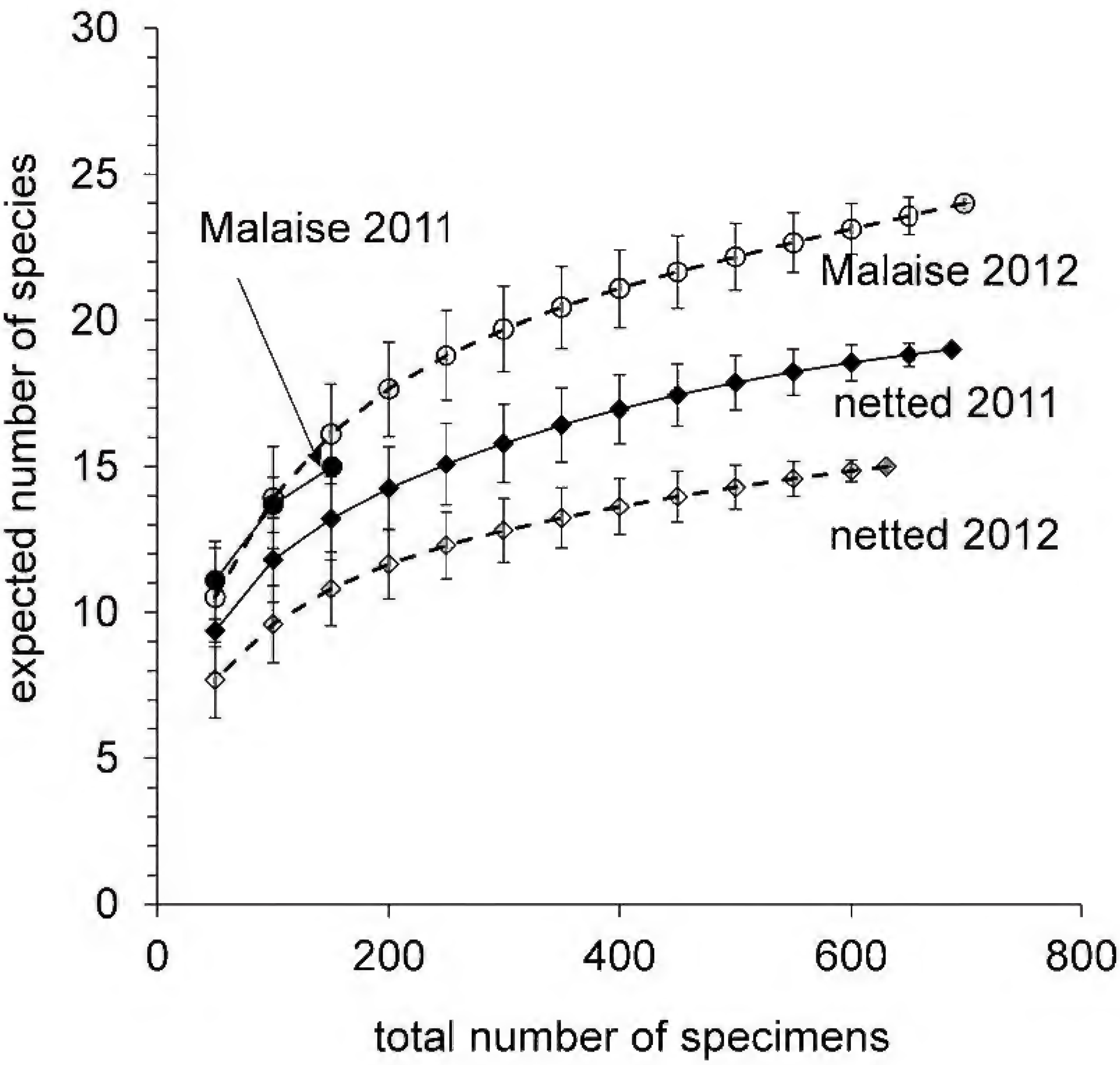

FIGURE 2. Rarefaction analysis of 2011 and 2012 data, separated by trapping method. Malaise traps (circles) and sweep netting (diamonds) in 2011 (closed) and 2012 (open). Error bars represent standard deviations.

Discussion

Comments on range extensions and new distributional locations are based on range maps from Teskey (1990), Thomas and Marshall (2009) and Thomas (2011). Because of the few intensive studies from northern Ontario we expected to add range records for many of the species we collected.

Our range map assessment underestimated by four the number of species we expected to catch, namely, 8 *Chrysops*, 16 *Hybomitra*, and two *Tabanus*; we caught 10 *Chrysops*, 18 *Hybomitra*, and two *Tabanus*. The lognormal prediction of 33 species was three more than what we found. Our survey did not include catches from August, and we expect there are more species in our study region that we did not manage to collect.

The two trapping methods collected different species (Table 1). Some species were abundant in both trapping methods, e.g. *C. excitans*, *H. epistates*, and *H. lurida*. *Hybomitra zonalis* was abundant in the Malaise collections, whereas *H. affinis* and *C. mitis* were abundant in the netted samples. Two species, *C. cuclux* and *H. nuda*, were absent from Malaise traps, and eight species, *C. ater*, *C. frigidus*, *C. niger*, *C. venus*, *C. zinzalus*, *H. frosti*, *H. hearlei*, and *T. vivax* were absent from the sweeps. When differences are examined within each year, the effect of trapping method becomes even more pronounced: nine species caught only by sweep netting and five only in Malaise traps in 2011; one species caught only by sweep netting and 10 only in Malaise traps in 2012 (Table 1, Fig 2). These results highlight the importance of collecting using a variety of methods in insect surveys to overcome catch biases. Other methods used to sample Tabanidae include larval collection (Philip 1928), chemical attractants (i.e., CO_2 or Octenol), baited traps such as the Nzi trap (Mihok *et al.* 2007), traps designed to act as visual cues for host seeking Tabanidae such as the unbaited Nzi traps (Mihok 2002), and Manitoba traps (Thorsteinson *et al.* 1964). Any trap designed to work using visual or olfactory cues for host seeking tabanids would produce high catches, most likely of host seeking females, but it is not known if these higher catches would result in proportionately more or different species. Larval collections do not rely on adult flight or host seeking, but are limited by the habitat that is searched (Philip 1928).

Generally, the most abundant species were caught over the longest period, with some exceptions. In 2011, *H. affinis* was the most abundant species (275 specimens), but was only caught during five sampling sessions (Table 2) whereas *H. arpadi* (33 specimens) and *H. lasiophthalma* (21 specimens), both relatively uncommon, were also captured during five of the sampling sessions. In 2012, *H affinis* (79 specimens) was caught over 9 sessions, but was less abundant than *H. lurida* (252 specimens), which was caught over 7 sessions (Tables I and II).

Acknowledgements

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PITFALLS AND PRESERVATIVES: A REVIEW

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Abstract

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An extensive review of the factors that affect the performance of arthropod pitfall traps is given. Liquid preservatives are discussed in a separate section because the choice affects the quality and composition of taxa collected in pitfalls.

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Introduction

Pitfall traps are a popular method for collecting ground beetles, spiders, ants and other epigeal arthropods (Westberg 1977; Niemelä *et al*. 1992; Bestelmeyer *et al*. 2000; Southwood & Henderson 2000; Phillips & Cobb 2005). While many shorter, general overviews exist (e.g., general techniques: Balogh 1958; Duffey 1972; Bestelmeyer *et al*. 2000; Southwood and Henderson 2000; Woodcock 2005; issues with pitfalls: Adis 1979), none have exhaustively examined the published literature recently. Herein we present such a review with the hope it will provide a sound base for those incorporating pitfall traps into research.

While the choice of preservative will affect the quality of specimens in any type of trap, it is a critical decision in pitfalls for several reasons. Chiefly, preservatives differentially attract and repel select arthropod taxa, which will affect the composition of taxa collected (Weeks & McIntyre 1997). Additionally, pitfalls are often set without covers in open fields, so lose more preservative through evaporation than other traps and are affected to a greater degree by rain and dilution by rainwater (Porter 2005). Therefore, we include a section detailing possible positives and negatives of preservatives used in pitfall traps.

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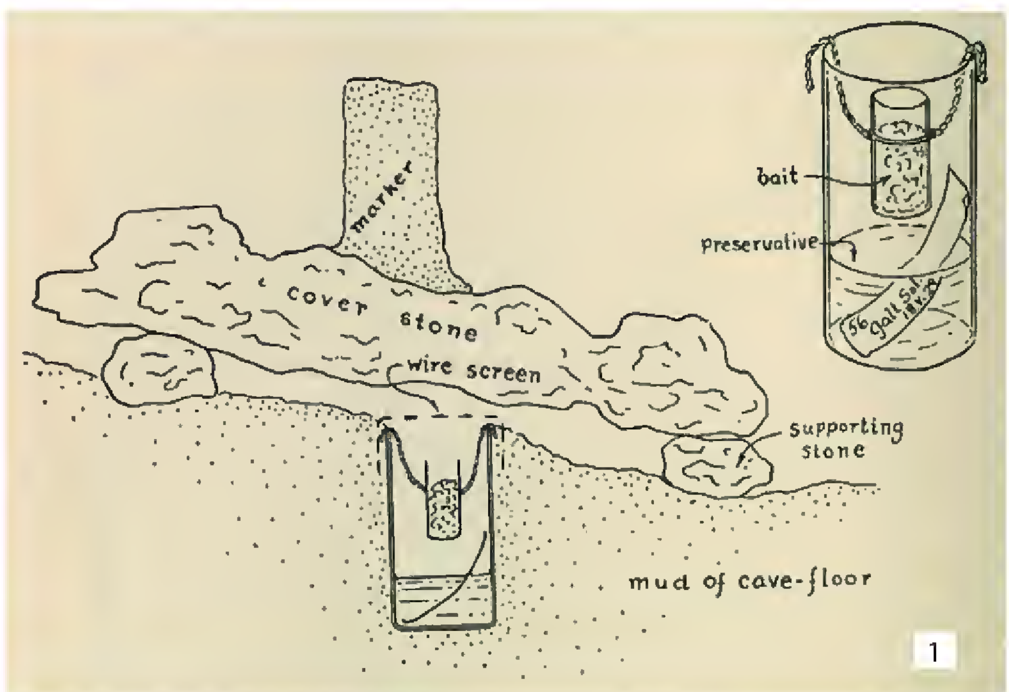
Pitfall Traps

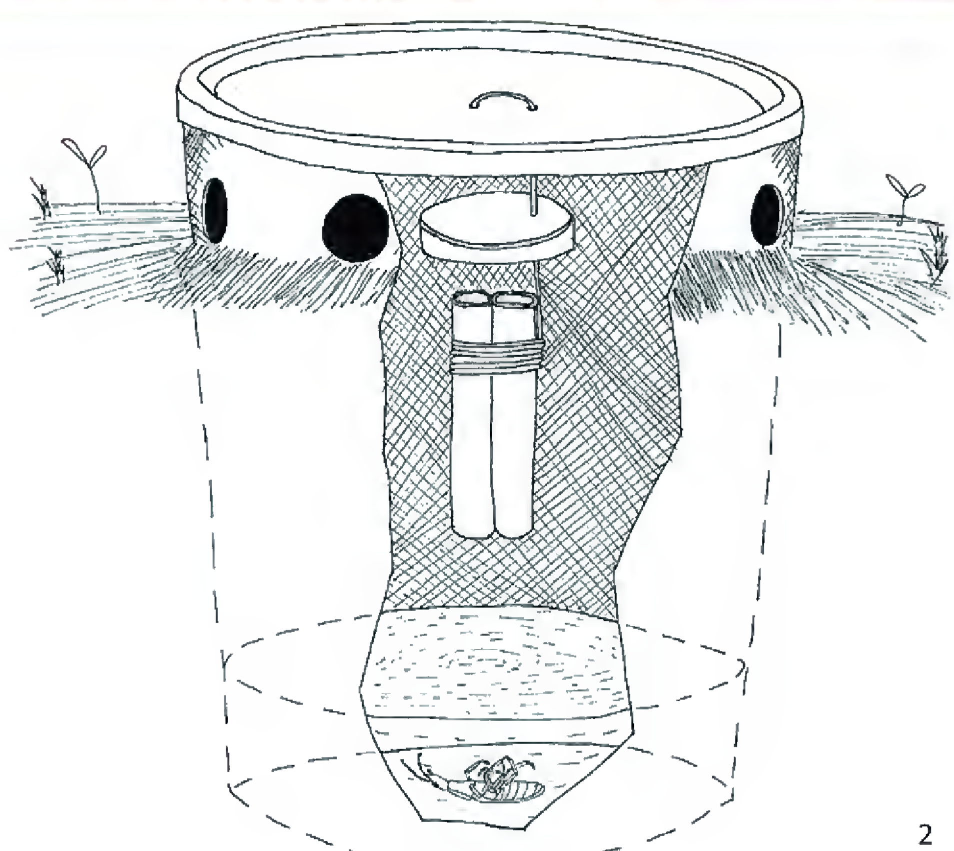
Pitfall traps were first described by Hertz (1927) and shortly thereafter by Barber (1931) (Fig. 1) for collecting cave-inhabiting insects. A pitfall trap is simple in design, consisting of a collecting container buried flush with the ground that passively collects epigeal organisms that accidentally fall into the trap. It may be constructed from any container large enough to hold the target organism, including a large bucket for reptiles or small mammals (Ellis 2013), small plastic cup for larger insects such as Carabidae and large Formicidae (Luff 1975; Abensperg-Traun & Steven 1995), or a glass test tube for small insects such as most Formicidae and small Carabidae (Luff 1968; Abensperg-Traun & Steven 1995). Pitfall traps are widely used in biodiversity surveys as they are cost-effective, ecologically sensitive, collect large numbers of arthropods (Gist & Crossley 1973; Ekschmitt *et al.* 1997; Southwood & Henderson 2000; Work *et al.* 2002), and collect nocturnal species missed by other methods (Törmälä 1982; Samways 1983; Donnelly & Gilmee 1985; Huusela-Veistola 1996).

Pitfall traps have been used to sample many arthropod groups, including Scorpionida (Tourtlotte 1974; Margules *et al.* 1994); Isopoda (Hamner *et al.* 1969; Hayes 1970; Paoletti & Hassall 1999; Hornung *et al.* 2007); Diplopoda (Van der Drift 1963; Kurnik 1988; Mesibov *et al.* 1995; Kime 1997; Snyder *et al.* 2006), Chilopoda (Kurnik 1988; Fründ 1990; Adis 1992; Shear & Peck 1992; Voigtlander 2003), and Symphyla (Adis 1992; Shear & Peck 1992; Clark & Greenslade 1996); Araneae (Duffey & Millidge 1954; Muma 1973; Uetz 1977; Corey & Taylor 1988; Bultman 1992; Koponen 1992; Bauchhenss 1995; Buddle *et al.* 2000); Acari (Zacharda 1993; Wickings 2007; Kłosin´ska *et al.* 2009; Mayoral & Barranco 2009; Wohltmann & Mąkol 2009; López-Campos & Vázquez-Rojas 2010; Clark 2013); Collembola (Joosse-van Damme 1965; Pedigo 1966; Budaeva 1993; Cole *et al.* 2001; Frampton *et al.* 2001); Coleoptera (Backlund & Marrone 1997; Simmons *et al.* 1998; Arbogast *et al.* 2000) including Carabidae (Anderson 1985; Kálás 1985; Cameron & Reeves 1990; Epstein & Kulman 1990; Togashi *et al.* 1990), Tenebrionidae (Ahearn 1971), Staphylinidae (Anderson 1985; Braman & Pendley 1993; Ekschmitt *et al.* 1997), Scarabaeoidea (Young 1981; Peck & Howden 1985; Martínez *et al.* 2009; Anlaş *et al.* 2011; Thakare *et al.* 2011), and certain Latridiidae (Hartley *et al.* 2007); Formicidae (Van der Drift 1963; Greenslade 1973; Anderson 1991; Abensperg-Traun & Steven 1995; Bestelmeyer *et al.* 2000); and even terrestrial Amphipoda (Craig 1973; Margules *et al.* 1994) and Decapoda(Williams *et al* 1985; Smith *et al.* 1991; Hamr & Richardson 1994; McGrath 1994; McIvor & Smith 1995). Of these taxonomic groups, ground-dwelling Araneae and Coleoptera have been the most studied (Westberg 1977).

Variations on the basic trap have been developed, including more elaborate traps for use under snow (Kronestedt 1968; Steigen 1973); live traps with a layer of gauze that keeps trapped organisms from drowning in rainwater (Duffey 1972); modifications that allow excess rainwater to drain before overflowing the trap (Duffey 1972; Porter 2005); integrated internal funnel and rain cap (Fichter 1941); collecting cup integrated into a larger structure with a base or ramp (Muma 1970); use of holes or slits in the side of a container so an integrated cap can be used (Fig. 2) (Nordlander 1987; Lemieux & Lindgren 1999); modifications to facilitate emptying (Rivard 1962), including automated devices for segregating trap catch over time (Williams 1958; Blumberg & Crossley, 1988; Buchholz

2009); designs to reduce mortality of vertebrate bycatch including floating shelters and wire mesh (Kogut & Padley 1997; Pearce *et al*. 2005); and inexpensive designs using commonly discarded household materials (Morril 1975; Clark & Bloom 1992). Other techniques, such as using an auger bit to drill placement holes for small diameter traps, and equipment, such as a device that can pull traps out of placement holes without kneeling or disturbing the surrounding soil, have been developed to make pitfall trapping easier (Vogt & Harsh





FIGURES 1–2. 1, Pitfall trap described by Barber for collecting cave-inhabiting insects. After Barber (1931). 2, Pitfall trap modified with entrances in the side of the collection cup, which discourages vertebrates from entering the trap and allows the use of an integrated rain cap. Modified from Nordlander (1987) with permission.

2003).

Barrier fences have been employed, either with a single pitfall situated in the middle of the fence or with pitfalls at the end of the fence (Fig. 3) (Haeck 1971; Meijer 1971; Reeves 1980; Durkis & Reeves 1982). Linear pitfall traps constructed from house gutters have been employed with success in certain situations, such as investigating the speed and timing of insect populations moving between habitats (Pamanes & Pienkowski 1965; Goulet 1974; Pausch *et al*. 1979).

Ramp traps collect arthropods similarly to pitfall traps, but rather than being sunk into the ground target taxa are directs upwards into the trap via ramps; this allows

FIGURES 3–4. 3, Pitfall traps (modified from Nordlander 1987) on either side of a barrier fence. 4, Ramp trap.

them to be employed where conventional pitfalls cannot, such as where digging is difficult (e.g., on rocks or in caves) or prohibited by law (Bouchard *et al.* 2000; Campbell *et al.* 2011). Bostanian *et al.* (1983) proposed the first ramp trap design, which is constructed from metal, making it rather bulky and expensive and biased towards large ground beetles. Bouchard *et al.* (2000) proposed a revised design that utilizes plastic sandwich containers and plastic ramps, rendering it light-weight and inexpensive (Fig. 4). Ramp traps have been successfully employed in caves (Campbell *et al.* 2011), areas polluted due to industrial mining (Babin-Fenske & Anand 2010), orchards (Smith *et al.* 2004), and vineyards (Goulet *et al.* 2004). Ramp traps capture a higher abundance and diversity of epigeal spiders than conventional pitfall traps, though when comparing other taxa (e.g., beetles) they collect a different species composition, thus making direct comparison between the trap types difficult or impossible (Pearce *et al.* 2005; Patrick & Hansen 2013). Additionally, ramp traps capture fewer vertebrates than conventional pitfall traps (Pearce *et al.* 2005).

Colored pan traps, sometimes referred to as water traps, are generally used to collect flying insects via visual response to color cues (e.g. yellow, blue, purple or red) (Kirk 1984; Aguiar & Sharkov 1997; Leong & Thorp 1999; Pucci 2008; Gollan *et al.* 2011). While pan traps are generally set on or above the ground, they may be sunk into it, effectively becoming pitfall traps that also attract and capture flying insects.

Issues with pitfall traps

Objections have been raised to the use of pitfall traps in ecological studies (Adis 1979; Majer 1997; Southwood & Henderson 2000) because they do not evenly catch different taxa for several reasons:

1. Different taxa react differently at the lip of the trap. Gerlach *et al.* (2009) found that millipedes show the most trap-avoidant behavior (20–60%) and carabids show the least (10–25%); overall they found an average of 28% of taxa that encountered a trap were caught, with a range of less than 5% (*Enantiulus nanus* (Latzel, 1884) (Julidae)) to 70% (*Pterostichus burmeisteri* Herr, 1838 (Carabidae)). Luff (1975) found approximately 75% of Carabidae that encounter the edge of a pitfall are collected. In mark-recapture studies, some species become trap-shy if they have been caught previously while other species do not (Benest 1989).

2. Activity level (Ekschmitt *et al.* 1997), which is affected by variables such as species-specific behavior (Greenslade 1964; Curtis 1980; Anderson 1991; Topping 1993; Spence & Niemelä 1994; Obrist & Duelli 1996); differences between gender and age (Hayes 1970; Benest 1989; Topping & Sunderland 1992; Thomas *et al.* 1998) including mate-searching (Tretzel 1954), post-copulatory dispersal of females (Merrett 1967) and searching for oviposition sites (Duffey 1956); weather (Williams 1940; Briggs 1961; Greenslade 1961; Juillet 1964; Ericson 1979; Drake 1994); vegetation (Deseo 1959; Greenslade 1964; Novák 1969; Baars 1979), habitat structure (Melbourne 1999; Melbourne *et al.* 1997; Thomas *et al.* 1998), and habitat type (Melbourne *et al.* 1997); size (Luff 1975; Thiele 1977; den Boer 1981; Franke *et al.* 1988) and speed (Braune 1974; Adis 1976); and hunger and prey density (Grüm 1971; Müller 1984; Henrik & Ekbom 1994), also affect the number of organisms trapped, both within and between taxa (Southwood, & Henderson 2000) and are more influential factors than population size (Briggs 1961) in determining trap catch.

3. Larger species are caught in significantly higher numbers than smaller species

(Carabidae: Franke *et al* 1988; Spence & Niemelä 1994). Several reasons have been suggested for this. Larger, faster beetles are successfully caught a higher percentage of the time than smaller, slower beetles (Braune 1974; Adis 1976) – though some authors have found size and speed do not affect the ability to be caught (Luff 1975; Halsall & Wratten 1988). Smaller beetles may escape more readily from traps because scratches and soil on trap walls may be enough to support their mass as they try to climb out whereas larger beetles fall (Spence & Niemelä 1994).

4. Species-specific morphology can affect escape ability; e.g., *Demetrias atricapillus* (L.) has adhesive setae on the underside of the tarsi that allow it to climb out of pitfalls more easily than other similarly sized carabids (Halsall & Wratten 1988).

5. Pitfall traps do not accurately reflect absolute density of the organisms sampled. This has been demonstrated in the field (Grüm 1959; Briggs 1961; Mitchell 1963; Marsh 1984; Topping & Sunderland 1992) and experimentally in a caged system (Lang 2000) – though caution should be exercised interpreting caged results as they may be skewed by "trap-happy" beetles that prefer dry pitfalls as refugia (Adis 1979, citing Thomas & Sleeper 1977) and may suffer from "Kreb's effect" (Mac Arthur 1984). However, it should also be noted that some studies have recorded 73–96% capture rates of marked beetles in caged systems (Bonkowska & Ryszkowski 1975; Dennison & Hodkinson 1984; Desender *et al.* 1985; Desender & Maelfair 1986; Clark *et al.* 1995; Holland & Smith 1999) and one study found no difference between population estimates of millipedes, spiders, and beetles based on hand collecting or pitfalls in a caged system (Gist & Crossley 1973), suggesting such systems may accurately reflect absolute density in certain situations with specific taxa.

In response to these criticisms, various calculations have been proposed to correct for the differences between taxa collected and true population density based on locomotory activity and motility range (Heydemann 1953; Tretzel 1955; Braune 1974; Thomas & Sleeper 1977; Kuschka *et al.* 1987; Stoyan & Kuschka 2001; see also Seifert 1990), though these have been rejected by others (Adis 1979; Müller 1984; Franke *et al.* 1988; Gerlach *et al.* 2009).

Additionally, it has been argued that samples pooled over an entire season correctly represent local species abundance as variations due to weather and other factors that affect activity level are averaged out (Baars 1979; den Boer 1986; Luff 1982). Results of other studies are conflicting, with some showing a large amount of variation between sampling periods in similar habitat when the sampling periods are short (Niemelä *et al.* 1986), and others showing that traps set for short periods caught all species accumulated by longer trapping periods (Niemelä *et al.* 1990; Borgelt & New 2006). In addition, much of the cited research has only examined carabids caught by pitfalls. When collecting other taxa, pitfalls may estimate absolute population density relatively well (ants: Andersen 1991; Vorster *et al.* 1992; Lindsey & Skinner 2001; cursorial spiders: Muma & Muma 1949; Duffey 1962; Huhta 1971; Uetz & Unzicker 1976; tenebrionids: Thomas & Sleeper 1977).

Certain ecological questions, such as comparing taxa along a successional gradient (Bultman & Uetz 1982) or between similar plots (Koivula *et al.* 1999), may be answered as taxa will be equally biased to pitfall traps along the gradient or between plots.

Pitfalls can be used to answer non-ecological questions, such as investigating the phenology (Maelfait & Baert 1975), seasonal and circadian activity (Williams 1959a, b; Williams 1962; Breymeyer 1966a, b; Doane & Dondale 1979), and lifespan

(Goulet 1974) of commonly collected taxa, estimating the timing of movement of epigeal species between habitats (Pamanes & Pienkowski 1965; Pausch *et al*. 1979), and estimating dispersal using mark-release-recapture methods (Ericson 1977; Best *et al*. 1981). They also can be employed in taxonomic surveys, though should be paired with other sampling techniques that complement the deficiencies of pitfalls (Majer 1997)

Pitfall trap design

If pitfall traps are to be employed, several considerations must be made as there are many factors that can affect the taxa collected.

<u>Effects of shape, size, and material of receptacle</u>. The shape of the trap affects the composition and number of taxa collected (Cheli & Corley 2010). Pitfalls may be straight-sided or round (Southwood & Henderson 2000), depending on the container used; however, round and straight-edged traps with the same perimeter length catch different numbers of specimens (Braune 1974; Luff 1975; Adis 1976; Spence & Niemelä 1994).

Different diameters of pitfall trap collect different taxa at different rates. When examining ants, larger diameter pitfalls catch more species, though differences are primarily due to differential capture rates of rare species (Abensperg-Traun & Steven 1995). Work *et al*. (2002) compared catch rates and species richness of Carabidae, Staphylinidae, and Araneae across five diameters (4.5, 6.5, 11, 15, and 20 cm) of pitfall traps; they found that, after standardizing circumference, small traps caught more small carabids and staphylinids and large traps caught more wolf spiders. Luff (1975) found that small traps (2.5 cm dia.) were the most efficient at catching small species of carabids, while large traps (10 cm dia.) caught relatively more large beetles; however, their small traps were made of glass and large traps made of metal, which probably had a confounding effect on the results. Brennan *et al*. (1999) found the largest and second largest traps (17.4 and 11.1 cm dia.) they tested caught the most diverse assemblage of species, though considered the smaller of the two traps more appropriate for sampling spiders as it may decrease the potential of capturing non-target species. One option when using larger traps is to add a funnel to the trap in order to increase trap retention (Vlijm *et al*. 1961).

Another aspect of size is the depth of the trap. Shallow (8 cm) and deeper (15 cm) pitfalls do not effect ant diversity capture (Pendola & New 2007), therefore, when targeting ants, shallow pitfalls are preferred as small vertebrates, such as skinks, may escape more easily from them, thus reducing vertebrate bycatch. However, this has only been demonstrated in ants and may not hold true for large insects, such as some carabids, which are bigger than some small vertebrates.

Pitfall traps used to collect insects have been constructed out of glass (Briggs 1961; Greenslade 1964; Borgelt & New 2006; Pendola & New 2007), plastic (Luff 1973; Morrill 1975; Clark & Blom 1992; Spence & Niemelä 1994), or metal (Ahearn 1971; Hinds & Rickard 1973; Clark & Blom 1992). Choice of material can affect the taxa sampled in live traps as escape rates differ. One study on carabids found 0% escape from glass traps, 4% escape per day from plastic traps, and 10% escape per day from metal traps (Luff 1975). Other studies have also found glass pitfalls retain more arthropods than plastic or metal (Vennila & Rajagopal 2000), though one found no difference between glass and plastic traps (Waage 1985). Similarly, Topping and Luff (1995) found plastic traps with rough surfaces caught fewer linyphiid spiders than similar traps with smooth surfaces.

Finally, color of the pitfall trap affects the taxa collected: white and yellow traps catch higher numbers of Apidae, Araneae, Carabidae, Diptera, and Formicidae, while brown and green traps catch higher numbers of Isopoda (Buchholz *et al*. 2010).

Effects of trap design, layout, and site selection. Some studies have found that covers do not affect the composition of arthropods trapped by pitfall traps (Work *et al*. 2002; Buchholz & Hanning 2009; Cheli & Corley 2010) while others have found they do (Briggs 1961; Baars 1979; Spence & Niemelä 1994). Some of this may be due to the material used as a cover. Man-made covers, such as metal or ceramic tile, are generally used. Suggestions have been made to use natural material such as bark or rock for covers (van der Berghe 1992), though this has not been systematically investigated.

Pitfall traps that have an integrated cap and circular entrances in the sidewall of the trap (first proposed by Nordlander 1987) caught 80% of the same common carabid species as conventional pitfalls in one study (Lemieux & Lindgren 1999), but otherwise have not been thoroughly investigated and compared to conventional traps.

Pitfall traps must be level with the soil surface as excessive inclination of the soil ringing the traps may direct some arthropods away from the trap (Heydemann 1953). Similarly, a plastic disc surrounding the trap will influence sample size (Adis 1976).

Subterranean pitfall traps have been employed to trap hypogaeic ants (Yamaguchi & Hasegawa 1996; Anderson & Brault 2010; Berghoff *et al*. 2003; Schmidt & Solar 2010), though these preform no better than conventional pitfalls (Pacheco & Vasconcelos 2012).

Use of a barrier fence consistently increases the number of ground beetles collected (Winder *et al*. 2001; Hansen & New 2005). However, the length of the fence influences trap catch (Durkis & Reeves 1982; Morrill *et al*. 1990), with longer fences catching higher diversity of families and species (Brennan *et al*. 2005), making it difficult to compare trap catch between studies. Location and number of the traps along the fence and fence material may also affect trap catch, though these variables have not been specifically investigated.

Spacing between traps is an important consideration as populations, especially of larger taxa such as carabids, can become locally depleted if traps are placed closely together; this can affect trap catch and skew results. Snider and Snider (1986) found no difference in trap catch between pitfalls spaced 0.5, 1, 2, and 4 meters apart. Similarly, Ward *et al*. (2001) found no difference in trap catch between pitfalls spaced 1, 5, and 10 meters apart. However, Digweed *et al*. (1995) found that carabid populations were depleted when pitfalls were placed 10 meters apart but not 25 meters; in addition, traps spaced at 10 meters had the most similar species assemblages and fewest rare species.

The optimum number of pitfall traps depends on the environment of the trapping site. As few as five traps aresufficient in an arid steppe environment (Cheli & Corley 2010), whereas ten to twenty pitfall traps effectively collected the majority of species in temperate areas (Formicidae: Santos *et al*. 2003; Coleoptera: Obrtel 1971; Isopoda Paoletti and Hassall 1999; Araneae: Niemelä *et al*. 1986), and at least twenty five are needed in tropical areas (Vennila & Rajagopal 1999). Various non-parametric estimators have been tested to estimate species richness based on as few as five traps per site (Brose 2002).

Finally, pitfall traps may not be the most efficient method for sampling epigeal arthropods in environments with rugged, steep slopes and a high density of rocks or roots in the soil where the traps are difficult to set or at high elevation where the mean body size of taxa is generally smaller, and thus more difficult to trap (Nyundo & Yarro 2007).

Additionally, some studies have found pitfalls trap more ants in drier areas and seasons (Delsinne *et al*. 2008; Nunes *et al*. 2011), though others have found annual rainfall has no effect (Delsinne *et al*. 2010).

Use of attractants in pitfall traps. The choice of preservative can affect the taxa collected in pitfall traps (Weeks & McIntyre 1997). For instance, bark beetles (Curculionidae: Scolytinae), certain Staphylinidae, and Nitidulidae are caught in higher numbers in pitfalls that use ethanol as the preservative (Drift 1963; Greenslade & Greenslade 1971). In one study, some Carabidae, especially *Bembidion*, were caught in higher numbers in ethylene glycol than water, though the effect varied by sex and time of year (Holopainen 1990, 1992); another study, however, found no difference between ethylene glycol and water when trapping four species of Diplopoda, one species of Chilopoda, and two species of Carabidae (Gerlach *et al*. 2009), suggesting that any effect is species dependent. Formaldehyde has been found to be repellant to Opiliones and Diplopodaand attractive to Carabidae and Staphylinidae (Luff 1968; Pekár 2002; Gerlach *et al*. 2009), though one study found no difference between water and formaldehyde when collecting Carabidae (Waage 1985). Differences have been found between commercially available antifreeze and diluted ethylene glycol (Koivula *et al*. 2003). Efficacy of preservatives can vary with trap size – one study found vinegar to be more effective in large traps but propylene glycol more effective in small traps (Koivula *et al*. 2003). Brine and an ethanol-glycerin mix have lower capture efficiency than other fluids such as pure water, ethanol-water, and ethylene glycol-water, possibly due to the high specific gravities of these fluids, which may allow captured arthropods to float and escape (Schmidt *et al*. 2006). Brine is also attractive to Lepidoptera (Cheli & Corley 2010). Additionally, attraction and repulsion to preservatives can vary due to sex (Adis 1976), season (Dethier 1947; Adis & Kramer 1975; Adis 1976), and environment (Koivula *et al*. 2003). Thus, careful consideration should thus be used in order to avoid or account for the influence of preservative on the taxa collected.

A drop of detergent is often used to break the surface tension of the preservative in wet pitfalls. This does not seem to affect the rate of capture of most arthropods, though Linyphiidae are caught in higher numbers (up to 1000%) in traps with detergent (Topping & Luff 1995; Pekár 2002), whereas Staphylinidae are caught in higher numbers in traps without detergent (Pekár 2002).

Some Coleoptera naturally aggregate using pheromones to locate conspecifics (Greenslade 1963; Wautier 1970, 1971; Ahearn 1971), which can affect trap catch distribution as the first specimen captured may artificially attract others to the same trap (Luff 1968; Thomas & Sleeper 1977; Luff 1986).

Digging-in effects have been recorded among Formicidae (Greenslade 1973), Carabidae (Digweed *et al*. 1995; Schirmel *et al*. 2010) and other Coleoptera (Schirmel *et al*. 2010), Collembola (Joosse-van Damme 1965; Joosse & Kapteijn 1968), Linyphiidae and other Aranaea (Topping & Luff 1995; Schirmel *et al*. 2010), and Isopoda (Schirmel *et al*. 2010). These effects consist of high capture of certain taxa immediately after pitfall traps are established followed by a subsequent decline. A variety of explanations – such as an increased level of CO2 (Collembola: Joosse & Kapteijn 1968), decreased barriers to movement (Carabidae: Greenslade 1964), increased number of prey that attract predators (Adis 1979), and decreasing number of foraging Formicidae workers (Romero & Jaffee 1989) – have been suggested, though no consensus has been reached. If digging-in effects

are to be avoided, it has been suggested either to place pitfalls inverted for one week before operating them as traps (Greenslade 1973; Schirmel *et al.* 2010) or to install a tube or second container in which the pitfall can be placed in order to avoid disturbing the soil when it is serviced (Schirmel *et al.* 2010). Alternatively, if the goal is to catch large numbers of arthropods without regard to comparing between-trap catch, traps may be serviced more frequently in order to take advantage of digging-in effects (Schirmel *et al.* 2010).

Disturbance of leaf litter and vegetation around the traps can cause increased catch of highly mobile taxa, such as Gryllidae (Sperber *et al.* 2007). Areas around active pitfalls should therefore not be visited unless the traps are being serviced. Alternatively, regularly scheduled visits to the trap area will increase the catch of certain mobile taxa, though care should be taken in designing and executing such visits in order to provoke the same disturbance between traps (Sperber *et al.* 2007).

If attraction is desired, baits can be used to purposely affect the taxa collected (Greenslade & Greenslade 1971). Dung and carrion can used to collect Scarabaeidae, Staphylinidae, Silphidae, Ptiliidae, Histeridae, Hydrophilidae, and Leiodidae. Carnivore and omnivore dung provide good results – with human dung being among the most effective and readily available – while herbivore dung is generally poor (Newton & Peck 1975). Meat, tuna, and honey can be used as baits for ants (Romero & Jaffee 1989). Though not intentional, previously trapped insects may begin to rot in traps in which the preservative is ineffective due to dilution from rain or large numbers of trapped insects, thus attracting carrion feeding taxa (Holland & Reynolds 2005). Vegetable oils have been shown to increase the catch of ants in the tropics (Pacheco & Vasconcelos 2012), especially army ants (Weissflog *et al.* 2000; Berghoff *et al.* 2002; Berghoff *et al.* 2003), although this has not been studied in temperate regions.

<u>Pests of pitfall traps</u>. Occasionally, traps will be regularly disturbed by mammals between collections. Van der Berge (1992) presented three situations with the possible culprits and associated solutions. For traps where the cup is still in the hole but pushed up "just enough so that the rim is no longer flush with the soil" he suggests moles or voles whose passage has been obstructed are to blame and moving the cup a short distance usually resolves the problem. When one or a few cups, but not the entire trap line, are completely out of the hole, spilled clean, but not chewed on he suggests squirrels are attempting to burry or dig up nuts. Unfortunately, "one is helpless against squirrel disturbance". The third case is when many, and often the whole line, of cups are out of the hole and chewed or mangled. This, he suggests, is the work of raccoons, opossums or deer that are interested in consuming the preservative. Raccoons are intelligent and will continue to harass a line of pitfall traps if they are reset, so it is best to abandon the line or add a distasteful substance to the preservative. If deer are molesting the traps, it is best to switch from a salt-based preservative which is probably drawing their attention.

Preservatives

Pitfall traps can be used to collect insects to be kept alive or killed in preservative. If live specimens are required, such as for rearing experiments (as is common in parasitengone mites to correlate life stages) or in cases where the taxon of interest is endangered, e.g.

the American burying beetle (*Nicrophorus americanus* (Olivier, 1790)), traps are run dry without preservative. In such cases, traps must be checked at least daily, and often more frequently, so captured individuals do not succumb to heat, desiccate, drown in accumulated rain water, or become predated on by other captured organisms (Mitchell 1963; Luff 1968; Weeks & McIntyre 1997; Bestelmeyer *et al*. 2000; Moreau *et al*. 2013).

When collecting specimens to be killed, the choice of trap preservative is an important consideration as it will affect the quality of specimens, cost of trap maintenance, and how frequently traps must be serviced. Many authors have investigated the preservation properties of different chemicals and solutions, which are summarized herein.

Ethylene glycol was once used as a preservative, especially in pitfall and pan traps, as it has low volatility compared to ethanol and other alcohols (Martin 1977), is relatively inexpensive, and is readily available as antifreeze. When used in the field it has been reported to not preserve internal organs well and causes specimens to deteriorate to the point of breaking when pinned (Aristophanous 2010), though other studies report sufficient preservation (Sasakawa 2007; Cheli & Corley 2010). Because ethylene glycol is toxic to vertebrates (Thrall e al. 1984) and is readily ingested due to its sweet taste (Grauer & Thrall 1982), its use has been discouraged (Hall 1991).

The addition of bitter agents, such as quinine, to ethylene glycol has been suggested as a way to deter vertebrates from drinking the fluid (Hall 1991). Quinine added to ethylene glycol, propylene glycol, and formalin has been shown to have no effect on the number of spiders caught in pitfall traps; in addition, it improves the preservation quality of specimens collected in ethylene glycol (Jud & Schmidt-Entling 2008). Alternatively, a red marking flag placed next to the trap may deter large vertebrates from investigating the trap and drinking the ethylene glycol (Cheli & Corley 2010).

An alternative to ethylene glycol but with similar characteristics is propylene glycol, which is sold as recreational vehicle and boat antifreeze. It also has low volatility and is inexpensive. Propylene glycol is nearly non-toxic as it is metabolized into constituents of the Krebb's cycle and extremely large quantities must be ingested over a short period of time before acute toxicity is reached (Yu 2007). In the field, propylene glycol preserves insects similarly to ethylene glycol (Jud & Schmidt-Engling 2008; Aristophanous 2010). However, Moreau *et al*. (2013) found no detectable difference in the quality of DNA preservation between propylene glycol and ethanol when undiluted chemicals were used in a lab setting. One reason for the difference between field and lab studies may be due to the fact that ethylene glycol and propylene glycol are hygroscopic; when humidity is moderate to high, both substances will absorb water from the air and dilute naturally (Aristophanous 2010).

Salt brine and saturated borax solution are inexpensive and easy to make as the constituent materials are readily available in grocery stores. The ability of these solutions to preserve insects is extremely poor, however, and not outweighed by cost-savings (Lemieux & Lindgren 1999; Sasakawa 2007; Aristophanous 2010) (though see Schmidt *et al*. 2006 for a counter opinion).

Carnoy's fixative (60% ethanol, 30% chloroform, 10% acetic acid) and white vinegar (10% acetic acid) do not preserve DNA and cause specimens to become brittle, though they generally keep the specimens from rotting (Sasakawa 2007; Aristophanous 2010; Moreau *et al*. 2013). If DNA extraction is not intended, these may be acceptable preservatives.

Methanol and chloroform do not preserve specimens in a way that allows DNA extraction and amplification (Post *et al.* 1993; Fukatsu 1999). In addition, chloroform is difficult to acquire, especially in the large quantities required for use as a trap preservative.

FAACC solution (formaldehyde 4%, acetic acid 5%, calcium chloride 1.3%) and 4% phosphate buffered formaldehyde (4%PBF) both preserve internal organs well, with 4%PBF being the superior of the two (Aristophanous 2010). However, specimens become excessively stiff and although DNA can be extracted from specimens preserved with formaldehyde solutions, DNA amplification is impossible with standard kits (such a Qiagen DNEasy) because formaldehyde causes DNA to cross-link with proteins (Schander & Halanych 2003). Protocols using prolonged extraction times (up to 7 days) (France & Kocher 1996; Chatigny 2000; Schander & Halanych 2003) and chemical agents (Johnson *et al.* 1995; Chatigny 2000) can be successful.

Amyl acetate is sometimes used in insect jars as the killing agent. This banana-smelling liquid keeps specimens relaxed, unlike other killing agents such as chloroform (Woodward 1951). It is commonly used as a water-removing solvent in industry and can be purchased through specialized suppliers. Amyl acetate has been used for preservation of anatomical dissections (Saunders & Rice 1944) and insects "may be kept stored almost indefinitely between cotton-wool impregnated with this agent" (Woodward 1951), though it has not been tested for DNA preservation (Nagy 2010). Additionally, it has not been tested as a preservative in pitfall traps, can be a skin irritant, and is probably attractive to some insect groups so other, more proven preservatives may be a better choice.

Ethanol is probably the most widely used preservative. It maintains the integrity of internal organs and allows DNA to be easily extracted and amplified (Gurdebeke & Maelfait 2002; Aristophanous 2010; Moreau *et al.* 2013). In the United States, price may be prohibitive for individuals who do not qualify for ethanol tax exemption; however, fuel ethanol has been shown to preserve specimens as well as pure ethanol, so this will provide an alternative source as fuel ethanol becomes more widespread (Szinwelski *et al.* 2012). In addition, ethanol is the most volatile commonly used preservative. In open containers such as pitfall traps ethanol can lose ¾ of its volume in fewer than 5 days (Aristophanous 2010). Depending on the trap location this may have implications on how often the traps must be serviced.

Isopropanol, commonly known as rubbing alcohol, is a cheap alternative to ethanol. Similar to ethanol, it preserves DNA well (Rake 1972), so it can be extracted with little difficulty. One drawback is that isopropanol often discolors specimens, which is a hindrance to identification and morphological studies involving color.

Acetone has shown promise as a preservative. It is relatively inexpensive and readily available as a paint solvent. DNA has been extracted and successfully amplified from acetone-preserved Copepods (Goetze & Jungbluth 2013), pea aphid (*Acyrthosiphon pisum* (Harris, 1776)) (Fukatsu 1999), and Zygoptera (Logan 1999). Additionally, acetone is used to preserve adult Odonata as it dissolves fat, dehydrates the specimen, and reduces decomposition of enzymatic color pigments (Abbott 2008).

Other preservatives require more testing as contradictory results have been reported. Fukatsu (1999) reported DNA amplification after specimens were stored in 2-propanol, ethyl acetate, and diethyl ether, though Post *et al.* (1993) and Reiss *et al.* (1995) reported poor results with 2-propanol and ethyl acetate, respectively.

Summary

Pitfall traps are often used to sample epigeal arthropods as they are inexpensive and easy to use. However, many factors influence the taxa so collected. Abiotic factors, such as weather, season, slope and aspect, degree of rockiness, and trap characteristics (color and material of the trap, diameter of the opening, spacing between traps, and number of traps at a site) affect the composition of collected taxa, often by affecting behavior of the target arthropods. Biotic factors affecting trap catch include species-specific factors (activity level, size, aggregation to conspecifics, and behavior at the edge of the trap), response to digging-in effects, and habitat structure, including the density of low-growing vegetation. The choice of preservative affects not only the level of preservation of specimens, but also the composition of specimens collected because various compounds differentially repel and attract different taxa. Taken together, these factors make comparisons between studies difficult.

While there have been calls to standardize pitfall trapping, the design employed in individual studies will continue to be based on the research question and materials available. An effort, however, should be made to report all of the factors that might influence the composition of specimens collected. While this may not be immediately useful, comparisons may be made in the future after further studies elucidate the effects various factors have upon trap catch.

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FIRST RECORDS OF *PTOSIMA WALSHII* (COLEOPTERA: BUPRESTIDAE) IN CANADA

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Ptosima Dejean, 1833 (Coleoptera: Buprestidae) (Fig. 1) contains 10 extant species worldwide (Bellamy 2008) with four species occurring in North American (Nelson 1978, Nelson et al. 2008). Nelson (1978) keyed and redescribed the North American species. Plant genera records for *Ptosima* spp. include *Crataegeus* (Rosaceae), *Cercis* (Fabaceae) and *Quercus* (Fagaceae) (Paiero et al. 2012). *Ptosima idolynae* Frost, 1923 and *P. laeta* Waterhouse,1882 are confined to south central North America, whereas *P. gibbicollis* (Say, 1823) and *P. walshii* LeConte, 1863 are widely distributed in eastern North America. Bright (1987) stated that *P. gibbicollis* "probably occurs in southern Ontario", but makes no mention of *P. walshii*. Paiero et al. (2012) mapped the distribution of *P. gibbicollis* as occurring in Ontario. They also mapped the host range of *P. walshii*, as a potential distributional range for the beetle, encompassing Manitoba, Ontario and Quebec, but no records were known for these Provinces. *Ptosima walshii* is considered "rarely collected" (MacRae 2006) and "infrequently encountered" (Paiero et al. 2012). Nelson (1978) stated that *P. walshii* had been reported from Illinois, Texas, Kansas and California, suggested that the California record was doubtful, and added new State distribution records for Iowa, Michigan, Minnesota, Mississippi, Missouri, Ohio and Wisconsin. Westcott (1991) provided compelling evidence that the California record was erroneous. Records for four states that border Canada—Michigan, Ohio, Wisconsin and Minnesota—are represented by a total of eight collections (Nelson 1978). The Michigan record was a single collection labeled "Ag. Coll. 20.V.1889" (Nelson 1978). Westcott (1991) reported a specimen from Oklahoma. Thus, the literature records confirmed by specimens include 11 States but no Provinces.

As part of a trapping study for the recently discovered European oak borer, *Agrilus sulcicollis* Lacordaire (Coleoptera: Buprestidae) in North America (Jendek and Grebennikov 2009, Haack et al. 2009), we collected numerous *P. walshii* adults in southwestern Ontario. These collections represent the first verified records of the species

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in Canada. Here we describe the circumstances of these collections.

Three woodlots (Table 1) selected for trapping in 2011, produced *P. walshii* adults. In each woodlot, three trap/lure combinations were deployed on oak trees: 1) unbaited sticky-band trap; 2) green prism trap baited with the green leaf volatile, 3-(*Z*)-hexenol (Synergy Semiochemicals, Inc., Burnaby, BC); and 3) green prism trap baited with a manuka oil/phoebe oil lure (Synergy Semiochemicals, Inc.). Three replicates of each trap type were established per site, for a total of nine traps per location. Traps were set up 19 May 2011 and monitored approximately every 6 or 7 days throughout the season until 18 August 2011. The Bickford Line and Courtright Line sites were sampled again in 2012 using unbaited green prism traps and sticky-band traps. Four replicates of each trap type were established per site for a total of 8 traps per location. Traps were set up on 10 to 12 May 2012 and monitored weekly until 19 July 2012. Three additional woodlots (Table 2), each with three sticky-band traps, also produced *P. walshii* adults. These additional traps were set up on 10 to 14 May 2012 and sampling was conducted once in mid-season (26 to 28 June) and again later in the season (18 to 19 July). The numbers of specimens of each sex collected at each location in each year are tabulated (Tables 1 and 2).

Males and females were collected from all three trap/lure types. Significantly more beetles (Fig. 2) of both sexes were collected on the green prism traps baited with the

FIGURE 1. Dorsal view of female of *Ptosima walshii* from Bickford Line site, Lambton, Co., Ontario (photograph by G.C. Jones).

TABLE 1. Number of adults of *Ptosima walshii* captured with green prism traps and sticky-band traps in woodlots in southwestern Ontario in 2011 and 2012.

			2011		2012	
Site	**Latitude**	**Longitude**	**Males**	**Females**	**Males**	**Females**
Bickford Line	42.7635°N	82.3096°W	6	11	11	22
Courtright Line	42.7987°N	82.2388°W	2	11	0	3
Thames Road	42.8518°N	81.7256°W	1	2	-	-
Total			9	24	11	25

TABLE 2. Number of adults of *Ptosima walshii* captured with sticky-band traps in additional woodlots sampled in southwestern Ontario in 2012.

Site	Latitude	Longitude	Males	Females
Hillsboro Road	43.0051°N	82.0946°W	1	2
Coldstream C.A.[1]	43.0210°N	81.4981°W	0	1
Ladysmith Road	42.8164°N	82.3946°W	0	1
Waterworks Road	42.8954°N	82.2575°W	0	2
Total			1	6

[1]C.A. = Conservation Authority

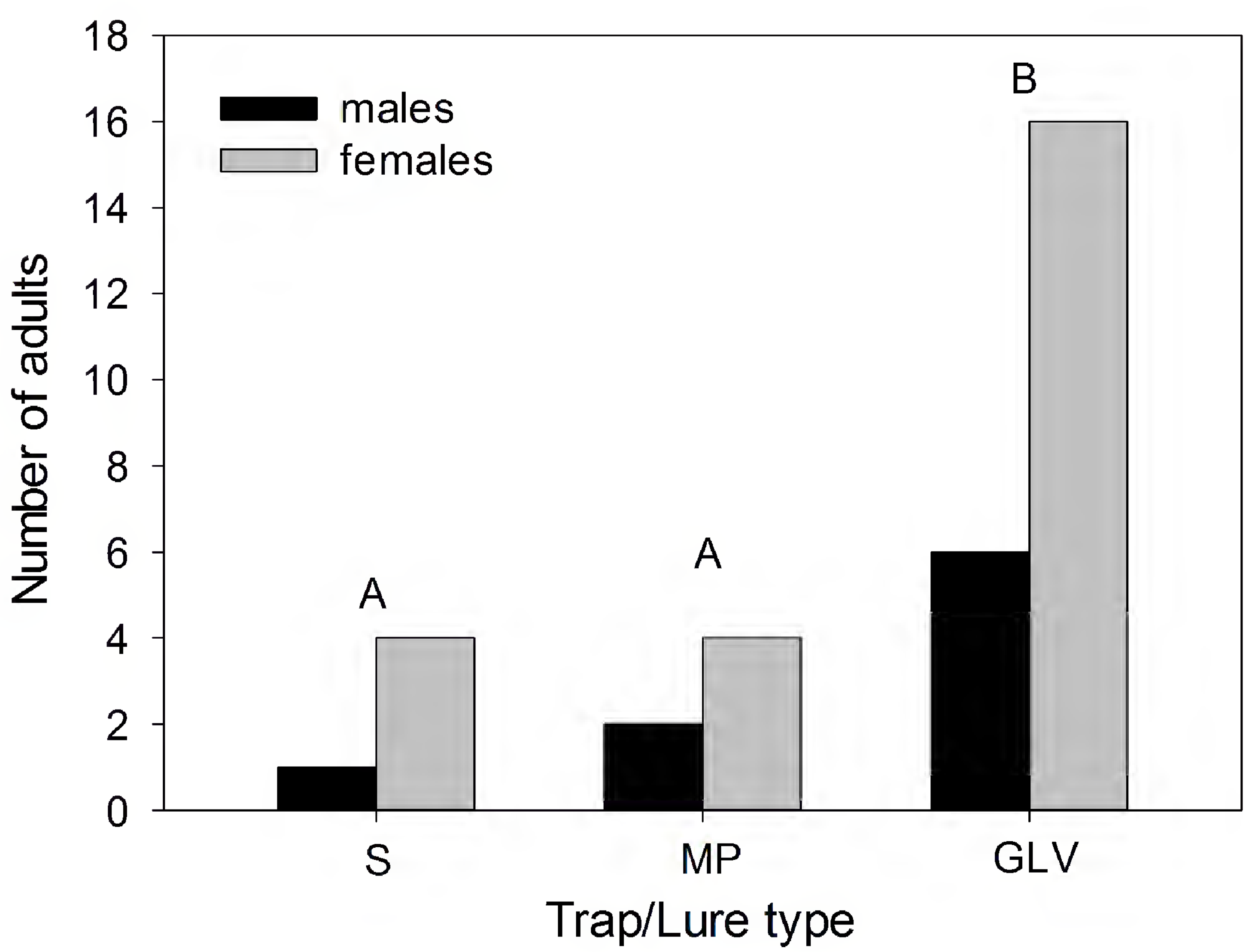

FIGURE 2. Number of males and females of *Ptosima walshii* captured in the three different trap/lure types (S= unbaited sticky-band traps, MP = green prism traps baited with manuka oil/phoebe oil; and GLV = green prism traps baited with 3-(Z)-hexenol) in four sites sampled in 2011 in southwestern Ontario. Different letters over the bars represent significant differences (G-test) between the total number of beetles (males + females) captured in each trap lure type.

3-(Z)-hexenol compared to the other trap/lure types (G-test; $\alpha = 0.05$, $G^2 = 24.247$) which suggests that this green leaf volatile is attractive to *P. walshii*. All adults of *P. walshii* were captured on either *Quercus macrocarpa* Michx. or *Q. alba*.L. Nelson (1978) listed the host of *P. walshii* as unknown. In a subsequent paper Nelson et al. (1981) reported that two adults had been collected by beating *Q. macrocarpa*. MacRae (2006) reported the first rearing of adults from *Q. macrocarpa*. The extensive use of this host tree for our trap placement serendipitously resulted in the capture of large numbers of *P. walshii*. The flight periods of the collected adults are shown in Fig. 3 for 2011 and 2012 excluding the additional sites. The early flight period of *P. walshii* observed in our study, especially in 2012, supports the assumption that the members of *Ptosima* overwinter as adults within pupal cells in the host tree (Nelson 1978).

Voucher specimens of *P. walshii* are deposited in the Great Lakes Forestry Centre Insect Collection (GLFC), the University of Guelph Insect Collection (DEBU) and the Canadian National Collection of Insects and Arachnids (CNCI).

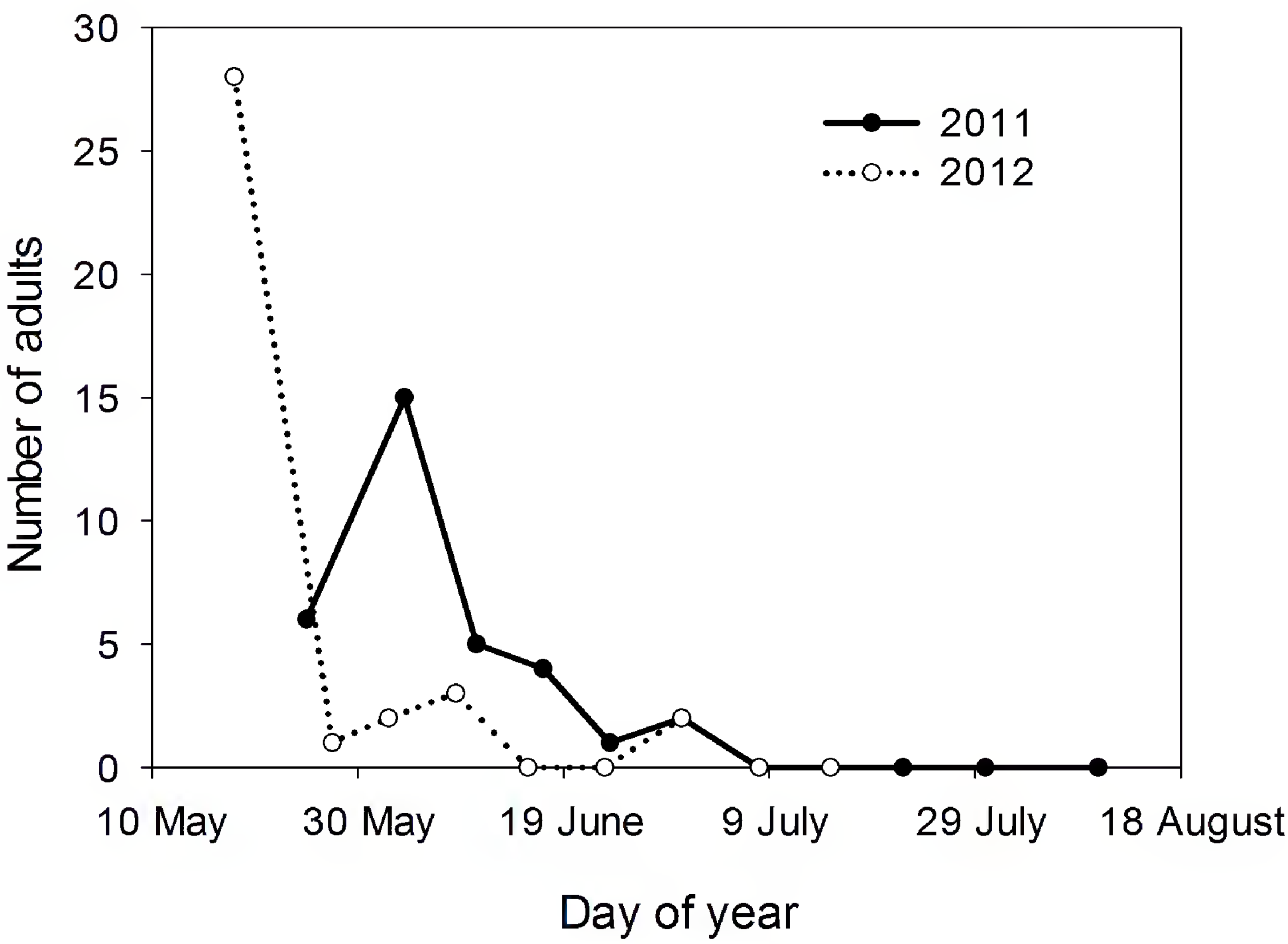

FIGURE 3. Total number of adults of *Ptosima walshii* (males + females) captured in each sample interval on all trap types (excluding additional woodlots in 2012), in 2011 and 2012 in southwestern Ontario, plotted in the middle of each sample interval.

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ECLOSION OF *PHYSOCEPHALA TIBIALIS* (SAY) (DIPTERA: CONOPIDAE) FROM A *BOMBUS* (APIDAE: HYMENOPTERA) HOST: A VIDEO RECORD

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Some members of Conopidae and other families of flies require development within hymenopteran hosts. Rearing of parasitized Apoidea provides valuable life history and ecological data but is rarely documented. Greater emphasis on gathering and analyzing rearing data is required. Analysis of a new video record of *Physocephala tibialis* (Say) reared from *Bombus impatiens* Cresson provides detailed evidence of the use of the ptilinum, mouthparts, and legs for eclosion within Conopidae. The previous literature on Conopidae/Apoidea rearing is reviewed.

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Introduction

Some species of Conopidae (Diptera) develop exclusively within hymenopteran hosts. Eggs are deposited inside adult bees or wasps. Following emergence from the egg, the larva grows, develops, and pupates entirely within the body of the host. Following death of the host and often after an overwintering period, the adult conopid ecloses from the host's corpse (Freeman 1966). While this behaviour is well noted within the conopid literature, careful rearing of parasitized hymenopteran hosts is only rarely documented.

Meijere (1904), Freeman (1966), and Smith (1959, 1966) summarized known host records and life histories for Conopidae, including many records of development within bee hosts. However, they did not distinguish between studies that relied on confirmed rearing records and studies that did not. Several studies tried to establish host records for Conopidae based exclusively on associations with host species (Rasmussen and Cameron 2004; Rocha-Filho et al. 2008) or on the discovery of eggs on pinned specimens (Stuckenberg 1963; Couri and Pont 2006; Couri and Barros 2010; Couri et al. 2013). These records must be considered

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tentative, as they do not confirm the successful development of Conopidae within a given host. While most confirmed rearing records are based on dead or obviously parasitized bees being held until parasitoids emerge, a few (Paxton et al. 1996; Polidori et al. 2005) were based on careful observation of host nests until parasitoids were seen to emerge.

Past rearing efforts have also produced other important observations on conopid life history. Schmid-Hempel et al. (1990) and Otterstatter et al. (2002) compared the relative parasitism rates and overlapping phenologies of competing parasitoid species. Knerer and Atwood (1967) reared one species of Conopidae, *Thecophora occidensis* (Walker), from six different species of halictine bees. They proposed a complex life history for *T. occidensis*, including phenological host switching. Müller (1994) observed self-burying behaviour in parasitized specimens of *Bombus terrestris*. Unfortunately, the conopid species reared out was never identified. Malfi et al. (2014) reared 39 specimens of *Physocephala tibialis* from three species of *Bombus*. They provided evidence of differential successful parasitism rates among the host species. Self-burying behaviour was also observed in parasitized bees, with a differential frequency of this behaviour depending on host species.

Several authors used rearing experiments to observe directly the process of eclosion from the host by Conopidae. Cumber (1949) reared *Physocephala rufipes* from *Bombus agrorum*, and noted that "the [conopid] adult emerges by pushing aside the anterior segments with its ptilinum." Polidori et al. (2005) observed an adult *Zodion cinereum* emerging from a ground nest of *Andrena agilissima* (Andrenidae) with the ptilinum "still being inflated rhythmically, indicating that they were freshly emerged adults." Koeniger et al. (2010) reared *Physocephala paralleliventris* from two different species of honey bees (*Apis cerana*, *A. koschevnikovi*) in Borneo. They observed an active period of walking for about fifteen minutes prior to inflation of the wings. They theorized that this active stage was necessary for conopids to emerge from the leaf litter in which the host bee was buried.

For other families of Diptera, video recordings of parasitoid emergence have been informative. Downing (1995) observed ptilinal expansion in *Amobia* (Sarcophagidae), which cleptoparasitizes mud-tube nesting wasps of *Trypoxylon* (Sphecidae). Strohm (2011) recorded cleptoparasitic members of Drosophilidae using ptilinal expansion to break out of the closed brood cells of their hymenopteran hosts.

The video recording presented here produced both life history data and behavioural observations for one species of Conopidae.

Materials and Methods

As part of ongoing research examining the impact of parasitism on bumble bees (Hymenoptera: Apidae: *Bombus*) in northern Virginia (Malfi and Roulston, 2014; Malfi et al. 2014), 445 foraging bumble bees were collected at Blandy Experimental Farm (Boyce, VA, USA, 36.09°N, 78.06°W) in June and July of 2012. These bees were maintained until death in the lab by housing bees in aquaria partially filled with soil and leaf litter and providing them sugar water *ad libitum* (for details, see Malfi et al. 2014). After death, bees were examined for the presence of a conopid parasite. Often this could be determined with minimal disturbance of the bee corpse as the conopid pupa frequently occupies the entire abdominal cavity of its host and a large, last-instar larva can be seen externally to move

within the bee. A total of 120 bumble bee workers belonging to three different species (*B. bimaculatus*, *B. griseocollis*, *B. impatiens*) were parasitized with conopid larvae. In September 2012, 94 conopid pupae harvested from these parasitized bees were placed in individual vials in a household refrigerator (~4°C) and left there until April, 2013,when they were placed on a lab bench in a room at 21.5° C and monitored for emergence. The emergence of one parasitoid was observed in progress. After the corpse of its host, a *Bombus impatiens* worker, began to move, one author (ADS) recorded the emergence process with a cell phone camera held up to the eyepiece of a dissecting microscope. Recording of emergence began at 8:50 AM on June 2nd, 2013, and continued for eight minutes, at which point the fly had completed its exit from the host.

Results

Thirty-nine of the conopid pupae emerged as adults and were identified as *Physocephala tibialis*. A 30-second edited version as well as the full eight-minute video may be viewed at (VIDEO). The edited video begins with the head of the conopid already visible, emerging from the ventral anterior of the bee abdomen. The ptilinum is seen fully inflated, with the antennae deflected to the ventral surface of the head (Figs. 1A, 1D). The mouthparts are displaced posteriorly, between the forelegs. At full inflation, the ptilinal sac appears to be equal in volume to the rest of the head. Following full inflation, the ptilinum deflates, but is still extruded (Figs. 1B, 1E). As the conopid pulls the rest of its body from the host, the ptilinum continues rhythmically inflating and deflating, though not to a volume equal to that visible as the head is first emerging. The conopid uses its legs and mouthparts as levers to pry itself from the host's body. When the conopid body is fully emerged from the host, it begins to walk around with the deflated ptilinal sac still visible. Throughout the eclosion process, it appears that the antennae, mouthparts, and legs are already sclerotized and dark. The sclerites of the head, thorax, and abdomen appear pale and unsclerotized. The wings are not inflated to any extent throughout the video. The adult *P. tibialis* is included with other voucher specimens that have been deposited in the University of Guelph Insect Collection, Guelph, ON.

Discussion

Schizophora (Diptera) have a membranous invagination in the head that is visible in adults only as a ptilinal fissure (Réaumur 1738; Becker 1882; Cumming and Wood 2009). The first description of the ptilinum in Calliphoridae (Réaumur 1738) included the suggestion that it was used to burst the puparium wall through expansion. Early research on the structure and function of the ptilinum (reviewed in Laing 1935; Atkins 1949) was limited to Calliphoridae and Drosophilidae. Strickland (1953) examined 150 species from over 40 schizophoran families and found that, in all cases, the ptilinum is lined with microscopic scales that are used to improve the puparium-bursting capabilities of the ptilinum. Ždárek et al. (1986) and Ždárek and Denlinger (1992) revealed pressure changes within the ptilinum and associated structures during eclosion and bodily inflation of the adult. Reid et al. (1987)

defined a series of phases, including ptilinal expansion in the 'extrication behaviour' of Sarcophagidae. Our observations suggest that Conopidae demonstrate the same patterns and behaviours of eclosion as those observed in Calliphoridae, Drosophilidae, and Sarcophagidae.

Strickland (1953) noted that the ptilinum of Conopidae is larger, thicker, and covered with more varied types of scales than that of any other fly family examined. His detailed drawings of *Physocephala furcillata* indicate that the base and length of the mouthparts as well as the ptilinum are covered with sclerotized scales. He contended that these scales assist with eclosion of the fly and subsequent digging from a subterranean location. The species we observed, *P. tibialis*, is morphologically similar to *P. furcillata* (Camras 1957). Malfi et al. (2014) demonstrated an induced digging behaviour in bees parasitized by *P. tibialis*. Our video demonstrates the use of the mouthparts and ptilinum as part of both eclosion and subsequent digging.

For most species of Conopidae, there are no host records confirmed through rearing. The known records for Conopidae reared from Apoidea are summarized here (Table 1). The seven genera listed represent only 11.9% of the approximately 59 extant genera and subgenera (Gibson and Skevington 2013; Gibson et al. 2013).

Using DNA barcoding approaches to associate larvae, pupae, or adults of parasitoids with each other and their host species (e.g., Smith et al. 2006, 2007) may add ecological and evolutionary data to rearing experiments. A search of the cytochrome oxidase *c* subunit I (COI) sequences available on GenBank (June 10, 2014) revealed 45 species of Conopidae,

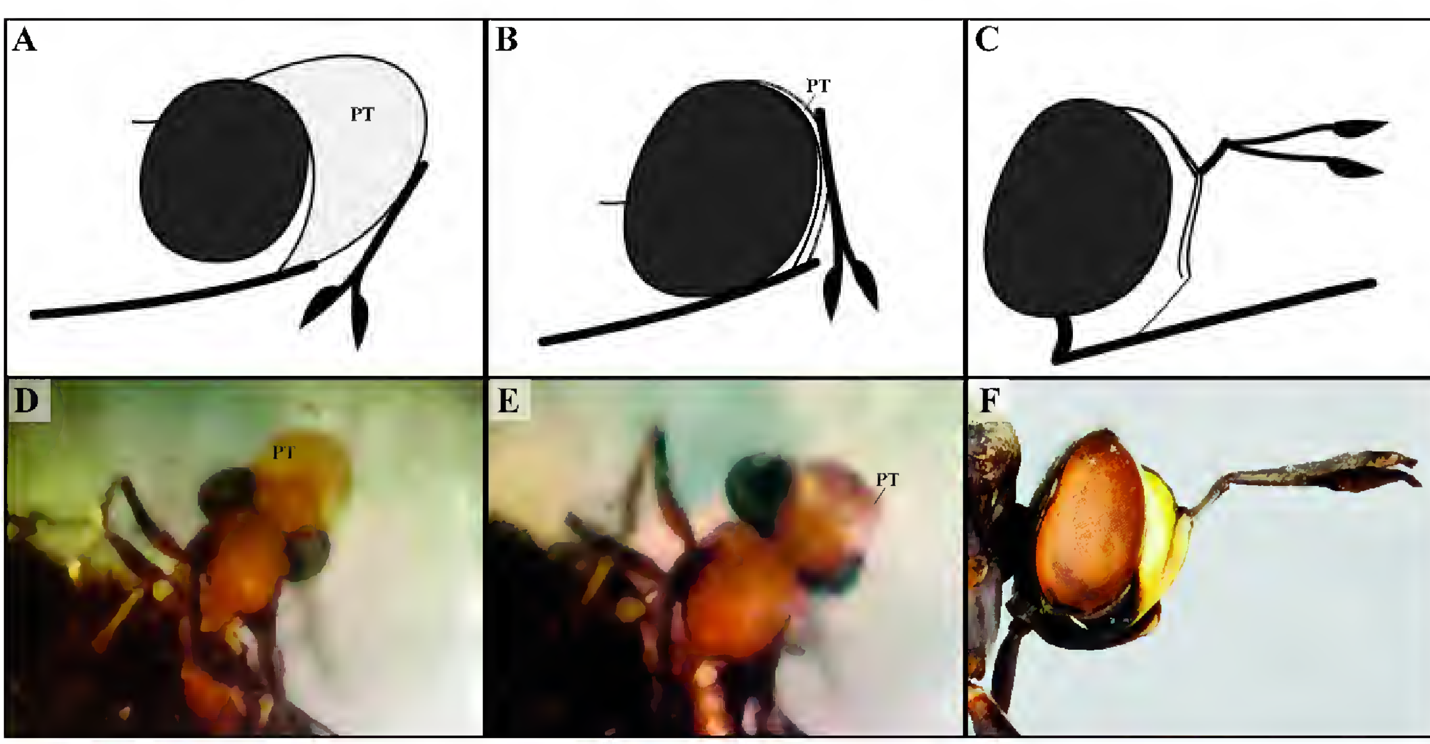

FIGURE 1. (A) *Physocephala tibialis.* Lateral view with ptilinum (PT) inflated. (B) Lateral view with ptilinum deflated. (C) Lateral view after sclerotization. (D) Dorsal view with ptilinum (PT) inflated. (E) Dorsal view with ptilinum deflated. (F) Lateral view after sclerotization (different specimen of same species). Photo supplied by T. Burt, Canadian National Collection of Insects. (D) and (E) are still images extracted from the VIDEO. Sclerotized adult head width for (D) and (E) = 3.69 mm. Line diagrams were created with Adobe Illustrator, based upon the video.

TABLE 1. Tabanidae species and number of specimens collected in 2011 and 2012 using Malaise traps and sweep netting, with abundance records.

Parasitoid	Host	Region	Reference
Dalmannia signata Chen	*Lasioglossum scitulum* (Smith)	Japan	Maeta and MacFarlane 1993
Myopa buccata (Linnaeus)	*Andrena japonica* (Smith)	Japan	Maeta and MacFarlane 1993
	A. scotica Perkins	Sweden	Paxton et al. 1996
M. rubida (Bigot)	*A. vierecki* Cockerell	California	MacSwain and Bohart 1947
M. testacea (Linnaeus)	*A. scotica* Perkins	Sweden	Paxton et al. 1996
Physocephala aurifrons Walker	*Centris analis* (Fabricius)	Brazil	Santos et al. 2008
P. bennetti Camras	*C. analis*	Brazil	Santos et al. 2008
	Xylocopa frontalis (Olivier), *X. submordax* Cockerell	Trinidad	Camras 1996
P. bimarginipennis Karsch	*X. carinata* Smith, *X. flavorufa* Degeer	Kenya, Uganda	Smith and Cunningham-Van Someren 1970
P. bipunctata (Macquart)	*Euglossa anodorhynchi* Nemésio	Brazil	Melo et al. 2008
	Centris analis (Fabricius)	Brazil	Santos et al. 2008
P. cayennensis Macquart	*C. analis*	Brazil	Santos et al. 2008
P. inhabilis (Walker)	*Megachile maculata* Smith	Brazil	Stuke and Cardoso 2013
	Centris analis	Brazil	Santos et al. 2008
P. furcillata (Williston)	*Bombus vagans* Smith	Ontario	MacFarlane and Pengelly 1975
P. marginata (Say)	*Apis mellifera* Linnaeus	Washington	Van Duzee 1934
	Bombus fervidus (Fabricius)	Ontario	MacFarlane and Pengelly 1975
	Megachile mendica Cresson	North Carolina	Krombein 1967
P. obscura Matsumura	*Megachile willughbiella* (Kirby)	Japan	Maeta 1997
	Bombus ardens Smith, *B. diversus* Smith	Japan	Maeta and MacFarlane 1993
P. paralleliventris Kröber	*Apis cerana* Fabricius, *A. koschevnikovi* Enderlein	Borneo	Koeniger et al. 2010
P. pusilla (Meigen)	*Megachile rotundata* Fabricius	Mongolia, France	Seidelmann 2005, Tasei 1975
P. rufipes (Fabricius)	*Bombus agrorum* Fabricius	England	Cumber 1949
	B. terrestris Linnaeus	???	Meijere 1904
	B. lapidarius (Linnaeus), *B. lucorum* (Linnaeus), *B. pascuorum* (Scopoli), *B. terrestris*	Switzerland	Schmid-Hempel and Schmid-Hempel 1988
P. rufithorax Kröber	*Centris analis*	Brazil	Santos et al. 2008
P. sagittaria (Say)	*Apis mellifera*	Washington	Van Duzee 1934
	Bremus auricomus Robertson	Illinois	Frison 1926

TABLE 1 continued...

P. spheniformis Camras	*Centris analis*	Brazil	Santos et al. 2008
P. soror Kröber	*Centris analis*	Brazil	Santos et al. 2008
P. texana (Williston)	*Apis mellifera*	Washington, Wyoming	Van Duzee 1934, Riedel and Shimanuki 1966
	Nomia melanderi Cockerell	Idaho	Foote and Gittins 1961
	Bombus bifarius Cresson, *B. californicus* Smith, *B. flavifrons* Cresson, *B. occidentalis* Greene	Alberta	Otterstatter et al. 2002
P. tibialis (Say)	*B. bimaculatus* Cresson, *B. griseocollis* DeGeer, *B. impatiens* Cresson	Virginia	Malfi et al. 2014
P. vittata (Fabricius)	*Megachile maritima* (Kirby)	Netherlands	Meijere 1904
P. wulpi Camras	*Xylocopa artifex* Smith, *X. augusti* Lepeletier, *X. splendidula* Lepeletier	Argentina	Stuke et al. 2011
Physocephala sp.	*Bombus* (9 spp.)	Massa-chussetts	Gillespie 2010
Physoconops fronto (Williston)	*Megachile perihirta* Cockerell	California	Bohart and MacSwain 1940
Sicus ferrugineus (Linnaeus)	*Bombus lucorum*, *B. pascuorum*, *B. terrestris*	Switzerland	Schmid-Hempel and Schmid-Hempel 1988
Thecophora occidensis (Walker)	*Lasioglossum forbesii* (Roberts), *L. laevissimus* (Smith), *L. lineatulum* (Crawford), *Halictus ligatus* Say, *H. rubicundus* (Christ), *Evylaeus cinctipes* (Provancher)	Ontario	Knerer and Atwood 1967
	Halictus confusus Smith	Indiana	Dolphin 1979
	H. confusus, *H. rubicundus*, *Lasioglossum cinctipes* (Provancher), *L. imitatus* (Smith), *L. lineatulum*, *L. forbesii*	Ontario	Smith 1966
Zodion cinereum (Fabricius)	*Andrena agilissima* (Scopoli)	Italy	Polidori et al. 2005
	Andrena prostomias Pérez	Japan	Maeta and MacFarlane 1993
Z. fulvifrons Say	*Apis mellifera*	South Dakota	Severin 1937
Z. obliquefasciatum (Macquart)	*Nomia melanderi*	???	Howell 1967
Z. vsevolodi Zimina	*Ceratina flavipes* Smith, *C. japonica* Cockerell, *C. megastigmata* Yasumatsu and Hirashima, *Chalicodoma spissula* (Cockerell), *Hylaeus thoracicus* Fabricius	Japan	Maeta and MacFarlane 1993

including three species of *Physocephala*. The use of DNA methods of identification, when target taxa have already been sequenced, greatly increases the value of studies that record immature parasites in hosts but, for methodological reasons, are unable to rear out adults (Gillespie 2010; Malfi and Roulston 2014).

Our study is an example of the added information about parasitoids that can be gained through careful rearing. Previous theories about the function of the ptilinum and the process of eclosion from the host have been strengthened with video evidence.

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NEW CANADIAN AND ONTARIO ORTHOPTEROID RECORDS, AND AN UPDATED CHECKLIST OF THE ORTHOPTERA OF ONTARIO

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The following seven orthopteroid taxa are recorded from Canada for the first time: *Anaxipha* species 1, *Cyrtoxipha gundlachi* Saussure, *Chloroscirtus forcipatus* (Brunner von Wattenwyl), *Neoconocephalus exiliscanorus* (Davis), *Camptonotus carolinensis* (Gerstaeker), *Scapteriscus borellii* Linnaeus, and *Melanoplus punctulatus griseus* (Thomas). One further species, *Neoconocephalus retusus* (Scudder) is recorded from Ontario for the first time. An updated checklist of the orthopteroids of Ontario is provided, along with notes on changes in nomenclature.

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Introduction

Vickery and Kevan (1985) and Vickery and Scudder (1987) reviewed and listed the orthopteroid species known from Canada and Alaska, including 141 species from Ontario. A further 15 species have been recorded from Ontario since then (Skevington *et al.* 2001, Marshall *et al.* 2004, Paiero *et al.* 2010) and we here add another eight species or subspecies, of which seven are also new Canadian records. Notes on several significant provincial range extensions also are given, including two species originally recorded from Ontario on bugguide.net. Voucher specimens examined here are deposited in the University of Guelph Insect Collection (DEBU), unless otherwise noted.

New Canadian records

Anaxipha species 1 (Figs 1, 2) (Gryllidae: Trigidoniinae)

This species, similar in appearance to the Florida endemic *Anaxipha calusa*

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Walker & Funk, is here recorded as new to Canada based on specimens found in 2013 in the Wings of Paradise Butterfly Conservatory, Cambridge, Ontario. Numerous individuals (including nymphs, adult males and females) were observed at that time, and discussions with the conservatory staff indicate that this *Anaxipha* species has been established there for some time. Two other Ontario butterfly houses were contacted to determine if this species had been introduced elsewhere in the province, but no further populations were reported. *Anaxipha* "species 1" is an undescribed species, probably the same as an unnamed species known to occur in Central America (Funk, pers. comm.; see also http://entnem.ifas.ufl.edu/walker/buzz/SM_AcalusaRelatives.pdf) and was probably accidentally introduced with shipments of butterfly pupae from Central America. If it is indeed this tropical species, it is unlikely to become established outdoors in natural habitats in Canada. Distinctive dark markings on the fore wings distinguish males (Fig. 1) of *Anaxipha* "species 1" from other

FIGURES 1–6. 1, *Anaxipha* species 1, ♂. 2, *Anaxipha* species 1, ♀. 3, *Cyrtoxipha gundlachi*. 4, *Chloroscirtus forcipatus*, ♀. 5, *Chloroscirtus forcipatus*, male terminalia (dorsal view). 6, *Camptonotus carolinensis*, ♀.

Ontario cricket species. The pale veins bordering infuscate cells make the females (Fig. 2) easy to separate from *A. exigua* (Say).
Specimen Records: ONTARIO: Wellington Co., Cambridge, Wings of Paradise Butterfly Conservatory, 43°27'7"N 80°22'2"W, in butterfly greenhouse, 20 April 2013, Paiero & Zinger (2 ♂♂); 3 July 2013, Paiero & Jackson (1 ♀, debu00367134); 22 May 2014, Paiero & Zinger (2 ♂♂; additional males and females from this collection were sent to Funk for further examination).

Cyrtoxipha gundlachi Saussure (Gryllidae: Trigidoniinae)

Cyrtoxipha gundlachi (Fig. 3), a species native to Florida and parts of the Caribbean (Eades *et al.* 2013), was found at the Cambridge Wings of Paradise Butterfly Conservatory and the Niagara Butterfly Conservatory. During visits to both sites, nymphs and adults were observed on foliage No native *Cyrtoxipha* species occur in Canada but *Cyrtoxipha columbiana*, which is similar in appearance to *C. gundlachi*, is found as far north as New Jersey (Walker 1969) and southern Ohio (Walker and Moore 2012). *Cyrtoxipha columbiana*, like *Anaxipha* species 1, may have been introduced with shipments of butterfly pupae, but it may have also been brought in with plants used within the greenhouses as both greenhouses have previously received plants from Florida.
Specimen Records: ONTARIO: Wellington Co., Cambridge, Wings of Paradise Butterfly Conservatory, 43°27'7"N 80°22'2"W, in butterfly greenhouse, August 2013, S.M. Paiero (3 ♂♂, 3 ♀♀, debu00367122-27); Niagara Reg., Niagara Falls, Niagara Butterfly Conservatory, 43°10'37"N 70°3'20"W, 22 June 2013, S.M. Paiero (2 ♀♀, debu00367135-36); same as previous except 8 August 2013 (1 ♂, 3 ♀♀, debu00367150-53); same as previous except 7 October 2013 (4 ♂♂, 2 ♀♀, debu00367128-133)

Neoconocephalus exiliscanorus (Davis) (Tettigoniidae: Conocephalinae)

Neoconocephalus exiliscanorus (the Slightly Musical Conehead) is here recorded from Canada for the first time, from a marsh adjacent to the Wheatley Provincial Park campgrounds. Although Vickery and Kevan (1985) indicate that *N. exiliscanorus* was expected to occur in Canada, the new Wheatley record is about 250 km north of the most northerly previous record. Its apparent preference for marsh habitat may be why this species had not previously been found. If *N. exiliscanorus* is established in extreme southern Ontario, additional populations may occur in similar habitats or along the shores of Lake Erie (e.g., Point Pelee).
Specimens examined: ONTARIO: Kent Co., Wheatley Provincial Park, 42°5'25"N 82°26'50"W, 10-11 August 2007, S.M. Paiero (2 ♂♂, debu00286872-73); same as previous except 7 September2007, S.M. Paiero (2 ♂♂, debu00291156-57).

Neoconocephalus retusus (Scudder) (Tettigoniidae: Conocephalinae)

Neoconocephalus retusus (the Round-tipped Conehead) is an eastern North American species here newly recorded from Ontario on the basis of observations and collections from the Haldimand-Norfolk region. Gartshore and Carson (pers. comm.) have heard this species singing at the same site in successive years, suggesting that the species is established in Ontario. If *N. retusus* was previously overlooked in Ontario it may be either due to its later emergence (Rehn & Hebard 1915) or because it has a restricted distribution in

the Norfolk area or because it was only recently established. Catling *et al.* (2009) previously recorded this species as new to Canada on the basis of specimens from Nova Scotia, but suggested that the species was adventitious rather than established.

Specimens examined: ONTARIO: Hald.-Norfolk Reg., Walshingham, ~5km SW, Pterophyla, 42°38'27"N 80°34'29"W, 31 August 2010, M. Gartshore, (1 ♂, debu00340774, one of several males heard singing by M. Gartshore and P. Carson), 27 September 2014, Gartshore & Carson (2 ♂♂).

Chloroscirtus forcipatus (Brunner von Wattenwyl) (Tettigoniidae: Phaneopterinae) (Fig. 4)

Chloroscirtus forcipatus is a Central American species that has established a population at the Niagara Butterfly Conservatory, likely being transported there in shipping materials. Feeding and oviposition by this exotic phytophagous species has caused significant damage to a wide variety of cultivated plants at the conservatory and discussions with staff indicate that it has been present there for many years.

Chloroscirtus forcipatus (Fig. 4) is superficially similar to the common bush katydids in the genus *Scudderia* but the hind femur has no spines on the lateral genicular lobe (Nickle 1992a), the eyes are more pronounced with the hind margin somewhat angled, the fore wings are more strongly microreticulate, the femora are shorter (only extending 2/3 length of fore wings; *Scudderia* extend to 4/5 or more), and the male cerci are distinct (Fig. 5). It will key out in Walker and Moore (2012) as *Turpilia rostrata* (Rehn & Hebard) although *T. rostrata* has spines on the lateral genicular lobe of the hind femur.

Specimens examined: ONTARIO: Niagara Reg., Niagara Falls, Niagara Butterfly Conservatory, 43°10'37"N 70°3'20"W, 22 June 2013, S.M. Paiero (2 ♀♀, debu00367137-38); 8 August 2013 (1 ♂, 2 ♀♀, debu00367154-56); 7 October 2013 (5 ♂♂, 3 ♀♀, debu00367160-67).

Camptonotus carolinensis (Gerstaeker) (Gryllacrididae)

Also known as the Carolina Leaf-rolling Cricket, *C. carolinensis* (Fig. 6) is here newly recorded from Canada on the basis of a specimen from Point Pelee National Park. Since *C. carolinensis* is the only representative of this group in North America, it represents a new family record for Canada. This predaceous species was previously known from Indiana to Florida (Walker and Moore 2012). *Camptonotus carolinensis* is easily overlooked as it is nocturnal and creates a leaf shelter in which to hide during the day, which might explain why it was not found earlier in Ontario.

Specimens examined: ONTARIO: Essex Co., Point Pelee Natl. Pk., West Beach, 41°59'0"N 82°32'50"W, wooded area, Malaise & pan traps, 10-23 September 1999, O. Lonsdale (1 ♂, debu00013739).

Melanoplus punctulatus griseus (Thomas) (Acrididae)

New records of *M. punctulatus griseus* from southern Ontario represent the first Canadian records of this subspecies, previously known from nearby Michigan (Vickery and Kevan 1985, Bland 2003). *Melanoplus punctulatus punctulatus* (Scudder), the other subspecies in Ontario, is widespread and extends east into Quebec. Bland (2003) reviewed the differences between the two subspecies.

Specimens examined: ONTARIO: Kent Co., Rondeau Prov. Pk., South Point Trail, nr. East

parking lot, oak savannah, 42°15'42"N 81°50'49"W, 10 October 2003, S.A. Marshall (1 ♂, 1 ♀, debu01134545-46); Essex Co., Point Pelee National Park, Visitor Centre, malaise & pans, O. Lonsdale, 11-18 August 2000 (2 ♂♂, debu01004046-47); same as previous except 5-26 September 2000 (2 ♂♂, debu01006370, debu01006376); same as previous except 27 August-5 September 2000 (1 ♂, debu01006403).

Scapteriscus borellii Linnaeus (Gryllotalpidae)

We identified one specimen of *S. borellii* from an Oakville bakery and it almost certainly represents an adventitious individual. *Scapteriscus borellii* is a Neotropical species (Nickel 1992b) that has become established in the southeastern U.S with its northernmost limit in North Carolina.

Specimen Examined: ONTARIO: Halton Reg., Oakville, found in a bakery, 13 October 1983, R. Ostrow (1 ♂, debu01076772).

Significant provincial range extensions

Meconema thalssinum (DeGeer) (Tettigoniidae: Meconematinae)

Meconema thalssinum (the Drumming Katydid) was originally recorded in Canada in 2004 (Marshall *et al.* 2004) on the basis of specimens from Harrow, and it has since been recorded in British Columbia as well (Cannings *et al.* 2007). This European species appears to have become widespread and established in Ontario from Windsor to Toronto, in both rural and urban environments.

Specimens examined: ONTARIO: York Reg., Toronto, 43°42'N 79°24'W, 8 July 2007, T. Careless, (1♂ 1♀, debu00296956-57); Toronto, prey of *Isodontia mexicana* (Hymenoptera: Sphecidae), July 2013, P.D. Careless (nymphs and adults; photograph); Toronto, 43°42'N 79°25'W, grass, sweep, 10 August 2007, A. Turko, (1♀, debu01031467); Essex Co., Harrow, 42°2'N 82°55'W, hand collection, 11 August 1997, M. Beaudoin (1♂ debu01031465); Durham Co., Darlington Prov. Pk., 43°52'17"N" 78°47'02"W", 11 September 2007, G. Vogg, (1♂, debu01031466); Kent Co., Wheatley Prov. Pk., 42°5'25"N 82°26'50"W, 22 July 2011, S.M. Paiero, (1♂ 1♀, debu00340211-12); Hald.-Norfolk Reg., Turkey Point Prov. Pk., site 2, 42°42'28"N" 80°20'29"W, savannah, at night , 4 August 2011, S.M. Paiero (1♂, debu01148938); Wellington Co., Guelph, Wellington Woods, 43°31'12"N 80°13'53"W, 11 August 2011, D.K.B. Cheung (1♀, debu00340228); Halton Reg. Oakville, nr. Hwy 25 & Burnhamthorpe Rd., August 2012, S.M. Paiero (1♂, debu00361466).

Neoxabea bipunctata (DeGeer) (Gryllidae)

This Carolinian species was originally recorded from Ontario on the basis of specimens from Essex County (Marshall et al. 2004). The more recent records presented here suggest that *N. bipunctata* now has a much more extensive range in southern Ontario.

Specimens examined: ONTARIO: Brant Co., Newport Forest, 30 July 2009, S.A. Marshall (1 nymph, photographed); Elgin Co., Springwater Forest, 3 October 2013, J. Allair (1♂, Allair 2013); Essex Co., Wheatley Prov. Pk., 1 Sep 2007, S.M. Paiero (1♀, debu00291096); Hald.-Norfolk Reg., Walshingham, ~5km SW, Pterophyla, 42°38'27"N 80°34'29"W,31 August 2013, at light sheet, Carson & Gartshore (2♂♂, 2♀♀; observed); same data as previous except 1 September 2013 (2♂♂, 2♀♀; photograph/observed); same

data as previous except 3 September 2013 (1♀; observed); Halton Reg., Oakville, nr. Hwy. 25 & Burnhamthorpe Rd., 29 August 2014, S.M. Paiero (1♂); Kent Co., Rondeau Prov. Pk., campground, 42°19'4"N 81°50'41"W, 25-26 September 2009, S.M. Paiero (1♀, debu00318141); Middlesex Co., London, Environmental Sciences Western Field Station, 43°4'29"N 81°20'13"W 13 September 2013, L. Des Marteaux (1♀, photograph).

Myrmecophila pergandei (Bruner) (Myrmecophilidae)

Although this species was first formally recorded from Canada during an insect survey of Ojibway Prairie, Windsor, Ontario (Paiero *et al.* 2010), the earliest Canadian collection of *M. pergandei* (the Eastern Ant Cricket) was from Ancaster, Ontario (Borer's Falls Conservation Area) in 2006, and we have also found it at Wheatley Provincial Park, Ontario. *Myrmecophila pergandei* is rarely encountered outside of ant colonies and most of the specimens we have observed were found in slave-maker ant colonies (*Formica* species) in Ancaster. Specimens from Wheatley Provincial Park and Ojibway Prairie Provincial Nature Reserve were found at night on the bark of trees, walking with foraging carpenter ants (*Camponotus* species).

Specimens examined: ONTARIO: Hamilton-Wentworth Reg., Dundas, Borer's Falls Conservation Area, in slave maker ant colony, in rotten fallen log, 24 May 2006, Umphrey, Marshall & Paiero (10 nymphs, debu00264402-264411); Kent Co., Wheatley Provincial Park, 42°5'25"N 82°26'50"W, on tree with carpenter ants at night, 22 July 2011, S.M. Paiero (1 ♂ 1 ♀, debu01154454-55); Essex Co., Ojibway Prairie, 42°15'51"N 83°4'30"W, 6 September 2007, S.M. Paiero (2 ♀♀, debu00291089-90)

Ectobius lapponicus (Linnaeus) (Ectobiidae)

This species, previously recorded in Canada from the Maritimes (Chandler 1992, Clements *et al.* 2013), is here recorded from Ontario for the first time although it appears to have been established in the province at least since 2006. Several specimens were collected during a 2014 "Bioblitz" in Toronto's Humber Valley and the species appears to be well established in the area. Several *Ectobius* nymphs collected during a 2013 "Bioblitz" in the nearby Rouge Valley are also likely to be *E. lapponicus*. Hoebeke and Carter (2010) gave features to separate this species from other introduced *Ectobius* in the northeast.

Specimen data: ONTARIO: Muskoka Distr., Gravenhurst, Muskoka Lake, 1 July 2006, on tree leaf, J.S. MacIvor, (1 ♂ debu01040861); same as previous except 2 July 2006 (1 ♀ debu01040862); Peel Reg., Mississauga, nr. Mississauga Rd. & Dundas Rd., 18 July 2008, S.M. Paiero, (1 ♂ debu00302523); York Reg., McMichael Canadian Art Gallery, 43°50'30"N 79°37'2"W, nymphs collected on gravel nr. lights, 24 May 2014, S.M. Paiero (5 ♂♂ 3 ♀♀ 3 nymphs); Halton Reg., Oakville, nr. Hwy.25 & Burnhamthorpe Rd., 25 June 2014, S.M. Paiero (1 ♂); same as previous except 1 July 2014 (1 ♂).

Ectobius lucidus (Hagenbach) (Ectobiidae)

Hoebeke and Carter (2010) reviewed the distribution of *Ectobius* in northeastern North America and gave characteristics to separate this species from other northeastern *Ectobius*, but did not record *E. lucidus* from Ontario. *Ectobius lucidus*, like *E. lapponicus*, was first recorded from Ontario (Orillia, 20 June 2008, "helmetinthebush", 1 ♀; Barrie, Ontario, 25 June 2005, "shemiles", 1 ♂) on the basis of images posted to and identified

on BugGuide.net. The additional specimen records below confirm the establishment of *E. lucidus* in Ontario, and further suggest that this species has been here since at least 1973. Specimen data: ONTARIO: Simcoe Co., Barrie, 2 August 1973, R.J. Hellewell, (1 ♂, debu01035451); Barrie, 11 July 2010, "vireo" (1 ♂, photo posted on bugguide.net); Midhurst, Neretra St., 2 July 2007, A. Brunke, (1 ♂, debu01035452); Simcoe Co., Georgian College, u.v. light, 20 July 1977, E.R. Fuller (1 ♂, ROM); Simcoe Co., S of Washago, mixed forest, 23 July 1992, L.D. Coote (1 ♂, ROM); Middlesex Co., London, Public Submission, 12 June 2001, T.A. Zowinski (1 ♂ 1 ♀).

Dubious record

Orocharis saltator Uhler (Gryllidae: Eneopterinae)

The University of Guelph Insect Collection has a specimen of *O. saltator* (the Jumping Bush Cricket) labelled as "Brant Co.?" without a collector or date. As no other Brant County exists in Canada or the United States, it is presumed that this locality refers to Ontario where it would be both a new species and subfamily record for Canada. The northern limit of the range of this species is close to southern Ontario but we have not included it in the checklist because the single record is doubtful.

Complete checklist of Ontario Orthoptera

Table 1 is a list of all 134 species and two additional subspecies of Orthoptera recorded from Ontario, including native species (125, including multiple subspecies), introduced species occurring outdoors (2), introduced species only found indoors (6), and adventitious species (7, including intercepted material). Table 2 is a list of the 36 other orthopteroids recorded from Ontario, including native species (6), introduced species occurring outdoors (8), introduced species only found indoors (8), adventitious species (9), and cultured species (5). Cultured species used in the pet trade were not treated as provincial records, as they are not known to occur in Ontario outside of captivity. Nomenclature follows Eades *et al.* (2013) for the Orthoptera, Beccaloni (2014) for the Blattodea, Deem (2014) for the Dermaptera, Otte *et al.* (2014) for the Mantodea, and Brock (2014) for the Phasmida.

Changes in status

Melanoplus differentialis differentialis (Thomas) was considered by Vickery and Scudder (1987) to be an introduced species. We consider it to be a native species as its occurrence in Ontario is close to the northern limit of its historical range. *Syrbula admirabilis* (Uhler) was also treated as an invasive by Vickery and Scudder (1987), but this species too has a historical range with Ontario being its northern limit. Although there are no recent Ontario records of *S. admirabilis*, historical records from southwestern Ontario are consistent with the overall range of the species, and it is here considered as native to the province. Similarly, *Parcoblatta caudelli* Hebard was treated by Vickery and Kevan (1987) as an adventitious species in Ontario but, based on the overall distribution of this species and its occurrence in Rondeau Provincial Park, we treat it as a native species. *Psinidia fenestralis fenestralis* (Audinet-Serville) was treated by Vickery and Scudder (1987) as "expected to occur in Ontario", but we have not included it in the checklist because we have yet to see a verifiable Ontario record of this species. *Schistocera americana* (Drury) was treated by Vickery and Scudder (1987) as an adventitious species not established in Ontario.

TABLE 1. Checklist of Ontario Orthoptera. (* denotes an introduced species occurring outdoors; $ denotes an introduced species only found indoors; ‡ denotes an intercepted or adventitious species).

ACRIDIDAE – Grasshoppers (52 species + 1 subspecies)
Acridinae (1 species)
Metaleptea brevicornis (Johansson)
Cyrtacanthacridinae (3 species)
Schistocerca alutacea (Harris)
Schistocerca americana (Drury)
‡*Schistocerca lineata* Scudder
Gomphocerinae (8 species)
Chloealtis abdominalis (Thomas)
Chloealtis conspersa Harris
Dichromorpha viridis (Scudder)
Orphulella pelidna (Burmeister)
Orphulella speciosa (Scudder)
Pseudochorthippus curtipennis curtipennis (Harris)
Pseudopomala brachyptera (Scudder)
Syrbula admirabilis (Uhler)
Oedipodinae (16 species)
Arphia conspersa Scudder
Arphia pseudonietana (Thomas)
Arphia sulphurea (Fabricius)
Camnula pellucida (Scudder)
Chortophaga viridifasciata (DeGeer)
Dissosteria carolina (Linnaeus)
Encoptolophus sordidus (Burmeister)
Pardalophora apiculata (Harris)
Spharagemon bolli Scudder
Spharagemon collare (Scudder)
Spharagemon marmorata marmorata (Harris)
Stethophyma gracile (Scudder)
Stethophyma lineata (Scudder)
Trimerotropis huronia E.M. Walker
Trimerotropis maritima (Harris)
Trimerotropis verruculata verruculata (Kirby)
Melanoplinae (23 species + 1 subspecies)
Booneacris glacialis canadensis (Walker)
Booneacris variegata (Scudder)
Dendrotettix quercus Packard
Melanoplus angustipennis (Dodge)
Melanoplus bivitattus (Say)
Melanoplus borealis borealis (Fieber)
Melanoplus bruneri Scudder
Melanoplus confusus (Scudder)
Melanoplus dawsoni (Scudder)
Melanoplus differentialis differentialis (Thomas)
Melanoplus eurycerus Hebard
Melanoplus fasciatus (F. Walker)
Melanoplus femurrubrum (DeGeer)
Melanoplus huroni Blatchley
Melanoplus islandicus Blatchley
Melanoplus keeleri luridus (Dodge)
Melanoplus mancus Smith
Melanoplus punctulatus griseus (Thomas)
Melanoplus punctulatus punctulatus (Scudder)
Melanoplus sanguinipes (Fabricius)
Melanoplus scudderi scudderi (Uhler)
Melanoplus stonei Rehn
Melanoplus walshii Scudder
Paroxya hoosieri (Blatchley)
Oxyinae (1 species)
‡*Oxya hyla intricata* (Stål)
ROMALEIDAE – Lubber Grasshoppers (1 species)
‡*Romalea microptera* (Beauvois)
TETRIGIDAE – Pygmy Grasshoppers (7 species)
Batrachideinae (1 species)
Tettigidea lateralis lateralis (Say)
Tetriginae (6 species)
Nomotettix cristatus cristatus (Scudder)
Paratettix cucullatus (Burmeister)
Tetrix arenosa angusta (Hancock)
Tetrix brunneri (Bolívar)
Tetrix ornata ornata (Say)
Tetrix subulata (Linnaeus)
TRIDACTYLIDAE – Pygmy Mole Crickets (3 species)
Ellipes gurneyi Günther
Ellipes minuta (Scudder)
Neotridactylus apicalis (Say)
MYRMECOPHILIDAE – Ant Crickets (1 species)
Myrmecophilus pergandei Bruner

TABLE 1 continued...

GRYLLIDAE – Crickets (23 species)
Gryllinae (4 species)
$*Acheta domesticus* (Linnaeus)
$*Gryllodes sigillatus* (F. Walker)
Gryllus pennsylvanicus Burmeister
Gryllus veletis (Alexander & Bigelow)
Nemobiinae (6 species)
Allonemobius allardi (Alexander & Thomas)
Allonemobius fasciatus fasciatus (DeGeer)
Allonemobius griseus griseus (E.M. Walker)
Allonemobius maculatus (Blatchley)
Eunemobius carolinus carolinus (Scudder)
Neonemobius palustris (Blatchley)
Oecanthinae (10 species)
Neoxabea bipunctata (DeGeer)
Oecanthus argentinus Saussure
Oecanthus exclamationis Davis
Oecanthus fultoni T.J. Walker
Oecanthus laricix T.J. Walker
Oecanthus latipennis Riley
Oecanthus nigricornis F. Walker
Oecanthus niveus (DeGeer)
Oecanthus pini Beutenmüller
Oecanthus quadripunctatus Beutenmüller
Trigonidiinae (3 species)
$*Anaxipha* species 1
Anaxipha exigua (Say)
$*Cyrtoxipha gundlachi* Saussure
GRYLLOTALPIDAE – Mole Crickets (2 species)
Neocurtilla hexadactyla (Perty)
‡*Scapteriscus borellii* Linnaeus
RHAPHIDIPHORIDAE – Camel Crickets (9 species + 1 subspecies)
Aemodogryllinae (1 species)
$*Diestrammena asynamora* (Adelung)
Ceuthophilinae (8 species + 1 subspecies)
Ceuthophilus brevipes Scudder
Ceuthophilus divergens Scudder
Ceuthophilus guttulosus guttulosus F. Walker
Ceuthophilus guttulosus thomasi Hubbell
Ceuthophilus latens Scudder
Ceuthophilus maculatus (Harris)
Ceuthophilus meridionalis Scudder
Ceuthophilus pallidipes E.M. Walker
Ceuthophilus uhleri Scudder
GRYLLACRIDIDAE – Leaf-rolling Crickets (1 species)
Camptonotus carolinensis (Gerstaeker)
TETTIGONIIDAE – Katydids (35 species)
Conocephalinae (20 species)
Conocephalus attenuatus (Scudder)
Conocephalus brevipennis (Scudder)
Conocephalus fasciatus (DeGeer)
Conocephalus nigropleurum (Bruner)
Conocephalus saltans (Scudder)
Conocephalus strictus (Scudder)
Neoconocephalus ensiger (Harris)
Neoconocephalus exiliscanorus (Davis)
Neoconocephalus lyristes (Rehn & Hebard)
Neoconocephalus retusus (Scudder)
Neoconocephalus robustus (Scudder)
‡*Neoconocephalus triops* (Linnaeus)
Orchelimum campestre Blatchley
Orchelimum concinnum Scudder
Orchelimum delicatum Bruner
Orchelimum gladiator Bruner
Orchelimum nigripes Scudder
Orchelimum silvaticum McNeill
Orchelimum voltanum McNeill
Orchelimum vulgare Harris
Meconematinae (1 species)
**Meconema thalassinum* (DeGeer)
Phaneopterinae (9 species)
Amblycorypha oblongifolia (DeGeer)
$*Chloroscirtus forcipatus* (Brunner von Wattenwyl, 1878)
Microcentrum rhombifolium (Saussure)
Scudderia curvicauda (DeGeer)
Scudderia fasciata Beutenmüller
Scudderia furcata furcata Brunner von Wattenwyl
Scudderia pistillata Brunner von Wattenwyl
Scudderia septentrionalis (Audinet-Serville)
Scudderia texensis Saussure & Pictet
Pseudophyllinae (1 species)

TABLE 1 continued...

Pterophylla camellifolia camellifolia (Fabricius)
Tettigoniinae (4 species)
Atlanticus davisi Rehn & Hebard
Atlanticus testaceus (Scudder)
* *Roeseliana roeselii roeselii* (Hagenbach)
Sphagniana sphagnorum (F. Walker)

TABLE 2. Checklist of Ontario's other orthopteroids (Blattodea, Dermaptera, Mantodea, Phasmatodea). (* denotes an introduced species occurring outdoors; $ denotes introduced species only found indoors; ‡ denotes an intercepted or adventitious species, % denotes a species that is reared in cultures in Ontario but not known to be established outside of these cultures).

BLATTODEA – Cockroaches (22 species) and Termites (3 species)
Blattidae (4 species)
$*Blatta orientalis* Linnaeus
$*Periplaneta americana* (Linnaeus)
$*Periplaneta australasiae* (Fabricius)
%*Shelfordella lateralis* (Walker)
Ectobiidae (5 species)
**Ectobius lapponicus* (Linnaeus)
**Ectobius lucidus* (Hagenbach)
‡*Nyctibora laevigata* (Palisot de Beauvois)
‡*Nyctibora noctivaga* Rehn
‡%*Symploce pallens* (Stephens)
Blatellidae (7 species)
$*Blatella germanica* Linnaeus
‡*Cariblatta sp. A*
Parcoblatta caudelli Hebard
Parcoblatta pennsylvanica (DeGeer)
Parcoblatta uhleriana (Saussure)
Parcoblatta virginica (Brunner von Wattenwyl)
$*Supella longipalpa* (Fabricius)
Blaberidae (6 species)
%*Blaberus discoidalis* Serville
%*Blaberus giganteus* (Linnaeus)
%*Blaptica dubia* (Serville)
%*Gromphadorhina portentosa* (Schaum)
‡*Panchlora nivea* (Linnaeus)
$*Pycnoscelis surinamensis* (Linnaeus)
(Infraorder ISOPTERA)
Kalotermitidae (1 species)
‡*Cryptotermes brevis* (F. Walker)
Rhinotermitidae (2 species)
**Reticulitermes flavipes* (Kollar)
‡*Reticulitermes virginicus* (Banks)

DERMAPTERA – Earwigs (8 species)
Anisolabididae (2 species)
**Anisolabis maritima* (Bonelli)
$*Euboriella annulipes* (Lucas)
Forficulidae (4 species)
‡*Chelidurella acanthopygina* (Gene)
Doru aculeatum (Scudder)
‡*Doru taeniatum* (Dohrn)
**Forficula auricularia* Linnaeus
Spongiphoridae (2 species)
**Labia minor* (Linnaeus)
$*Marava arachidis* (Yersin)

MANTODEA – Praying Mantids (2 species)
Mantidae (2 species)
**Mantis religiosa* Linnaeus
**Tenodera sinensis* Saussure

PHASMATODEA – Walkingsticks (1 species)
Heteronemiidae (1 species)
Diapheromera femorata (Say)

Since *S. americana* has a historical eastern North American range within flying distance of Canada, we consider it to be a native, albeit migrant, species. *Pycnoscelis surinamensis* was also recorded by Vickery and Kevan (1987) as an adventitious species in Ontario but this species is now well established in many greenhouses.

Nomenclatural changes

Nomenclatural changes since Vickery and Scudder (1987) are summarized below.

Acrididae

Schistocerca emarginata (Scudder) is now treated as a synonym of *S. lineata* Scudder (Song 2004).

Stethophyma was previously treated within the Acrididinae (Vickery and Kevan 1985) and Oedopodinae (=Locustinae, Vickery and Scudder 1987); it is now treated in the Oedopodinae (Chapco and Contreras 2007).

Chorthippus curtipennis curtipennis (Harris) is now treated as *Pseudochorthippus curtipennis curtipennis* (Harris) (Défaut 2012).

Arphia pseudonietana pseudonietana (Thomas) is now treated as *A. pseudonietana*; no subspecies are recognized (Otte 1984).

Orphulella pelidna pelidna (Burmeister) is now treated as *O. pelidna;* subspecies are no longer recognized (Eades *et al.* 2013).

Spharagemon bolli bolli Scudder is now treated as *S. bolli* Scudder; subspecies are no longer recognized (Otte 1984).

Trimerotropis verruculata (Kirby) is now treated as *Trimerotropis verruculata verruculata* (Kirby) because an additional subspecies is now recognized (*T. verruculata suffusa*; Eades *et al.* 2013).

Trimerotropis maritima interior Walker is now treated as *T. maritima* (Harris); subspecies are no longer recognized (Otte 1984).

Melanoplus viridipes eurycercus Hebard is now treated as a separate species, *M. eurycercus* Hebard (Otte 2002).

Melanoplus femurrubrum femurrumbrum (DeGeer) is now treated as *M. femurrubrum*; subspecies are no longer recognized (Eades *et al.* 2013).

Romaleidae

Romalea guttata (Houttuyn) is now treated as *R. microptera* (Beauvois) (the Eastern Lubber Grasshopper); it is apparently occasionally carried north from the southeastern USA in plant shipments or as bait by fishermen, but is not established in Ontario.

Tridactylidae

Ellipes minutus minutus (Scudder) is now treated as *E. minuta* (Scudder) (spelling corrected); subspecies are no longer recognized (Eades *et al.* 2013).

Tetrigidae

Tettigidea lateralis (Say) is now treated as *T. lateralis lateralis* (Say) as another subspecies is recognized (Rehn and Grant 1958). Vickery and Kevan (1985) recognized the

subspecies but it was omitted in Vickery and Scudder (1987).

Gryllidae
The Tropical House Cricket (*Gryllodes sigillatus* (F. Walker)) was previously confused with *G. supplicans* (F. Walker) (Otte 2006). *G. sigillatus* is occasionally brought into Canada as cultures and is commercially available as reptile food, but is not known to be established outside of cultures.

Rhaphidophoridae
Tachycines asynamorus Adelung is now treated as *Diestrammena* (*Tachycines*) *asynamorus* (Adelung); *Tachycines* has been lowered to subgeneric rank (Sugimoto 2002).

Tettigoniidae
Atlanticus davisi Rehn & Hebard was previously treated by some authors as a synonym of *A. monticola* Davis but its species status is here retained, following Eades *et al.* (2013). *Roeseliana roeselii* (Hagenbach) was placed in *Roeseliana* by Zeuner (1941) but has since been treated by most authors as belonging to *Metrioptera* or *Sphagiana*. Massa & Fontana (2011) reinstated *Roeseliana* as a valid genus. Multiple subspecies are also recognized and *R. roeselii roeselli* is the subspecies present in North America (Eades *et al.* 2013). *Pterophylla camellifolia* (Fabricius) is now treated as *P. camellifolia camellifolia* as several subspecies are recognized (Alexander 1968, Walker and Moore 2012).

Mantidae
Tenodera sinensis Saussure was originally treated as a subspecies of *T. aridifolia* Stoll; Ehrmann (2002) treated it as a separate species.

Discussion

Although the orthopteroids are relatively well studied in North America, further Canadian records are to be expected. Several species so far unknown from Canada have known ranges that almost reach the border, especially near southern Ontario. For example, *Oecanthus forbesi* Titus, morphologically indistinguishable from *O. nigricornis* F. Walker, likely occurs in southwestern Ontario based on range maps (Walker and Moore 2012) but has not yet been formally recorded from Canada. This species would be most effectively sought out on the basis of its song. Other species currently known from nearby localities just south of the border, such as *Phyllopaplus pulchellus* Uhler or *Trachyrhachys kiowa* (Thomas) are likely candidate additions to our fauna in response to a warming climate, and might already occur here as undocumented populations. There also remains some possibility of adding previously undescribed species to the provincial fauna. Several *Anaxipha*, for example, have only recently been described from the eastern USA (Walker and Funk 2014) and some of these may yet be discovered in southwestern Ontario. There is also a high probability of continued discoveries of newly introduced exotic species. Although most of the recently discovered introductions are of tropical species that are unable to survive

outside during the winter in Canada, several exotic orthopteroids (*Meconema thalassinum*, *Roeseliana roeselii roeselii, Ectobius lapponicus* and *E. lucidus*) have become successfully established in Ontario. And while the presence of *Anaxipha* species 1 is surprising, finding an undescribed tropical species in temperate climates is not unprecedented. Weissman *et al.* (2012) found a previously undescribed *Gryllus* species (*G. locorojo* Weissman & Gray, also known as the "crazy red"), apparently native to South America, being used as a feeder cricket in parts of Denmark, England and the United States.

The ongoing task of documenting the presence and ranges of orthopteroids (and other arthropods) in Canada is expedited by keys and photographic guides that enable amateurs and biologists without entomological training to recognize species. Vickery and Kevan (1985; available through the Entomological Society of Canada's website) remains an important resource although can be difficult to use without access to an extensive reference collection. Bland (2003), Walker and Moore (2012) and Kirk and Bomar (2005) are more extensively illustrated guides that cover parts of Ontario's fauna and supplement the keys given in Vickery and Kevan (1985). Correctly identified photos of most Canadian orthopteroid species can be found on bugguide.net (http://bugguide.net/node/view/15740) and Walker (2014) provided keys and songs to the Gryllidae and Tettigoniidae of North America.

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